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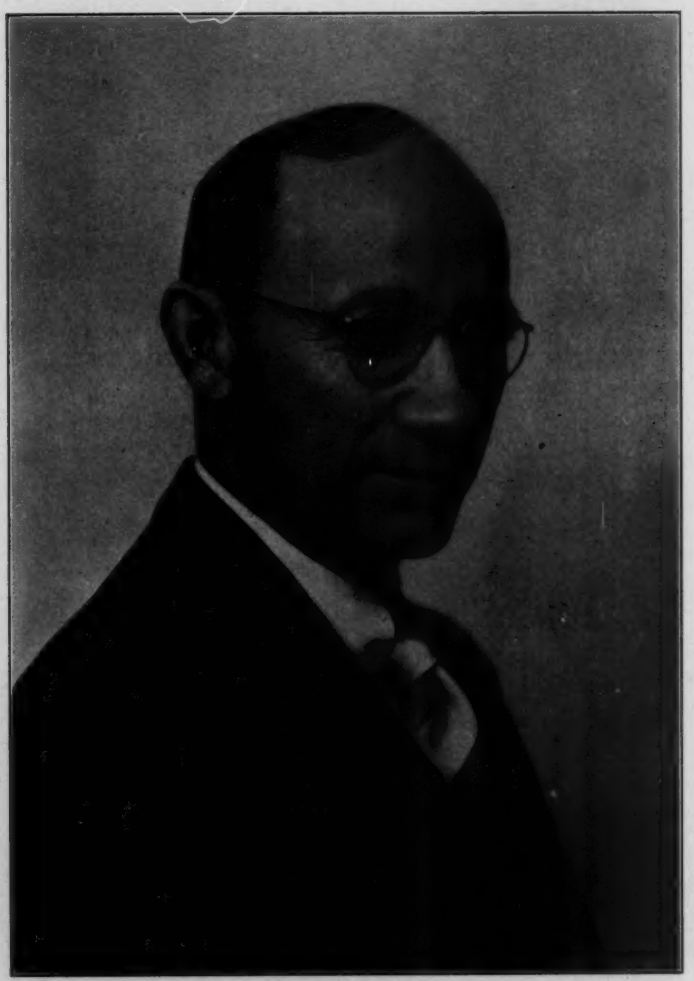

LEE ORAS OVERHOLTS

June 23, 1890–Nov. 10, 1946

MYCOLOGIA

OFFICIAL ORGAN OF THE MYCOLOGICAL SOCIETY OF AMERICA

VOL. XL JANUARY–FEBRUARY, 1948 NO. 1

LEE ORAS OVERHOLTS

FRANK D. KERN

In the death of Lee Oras Overholts at State College, Pennsylvania, November 10, 1946, mycology has lost one of its most distinguished students. His last illness had kept him away from work only for a week. For the last five years his health was impaired but except for short periods he had kept tenaciously at his work. All his life he was a prodigious worker, not sparing himself in health or in illness. Nights, Sundays, holidays and vacations were occasions for furthering the work so well begun during regular days and hours. He was always extravagant with his energy. His extensive collections of specimens, photographs and notes which he has left us are inadequate consolation for his passing but they are evidence of much rich living even though he died at the untimely age of fifty-six years.

Lee Oras Overholts was born June 23, 1890, in Camden, Ohio. He was first married to Flora May Conarroe who died June 7, 1944, and later he was married to Marie Knauz who survives with his four children: Mrs. Charles Rick, of Davis, California; Benjamin and Robert Overholts and Mrs. Eugene Zierdt, all of State College, Pennsylvania.

He attended Miami University, Oxford, Ohio, graduating in 1912 with the A.B. degree. In college he came under the influence of Professor Bruce Fink, an able and inspiring botanical teacher. His early aptitude for scientific work is evidenced by the fact that Professor Fink recommended him during the spring of his senior

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year (1912) to carry on Arthur's culture work with the rusts. It was there that I first met him and my belief that here was a man of exceptional promise prompted me to offer him in 1915 a position on the staff of the department of botany of the Pennsylvania State College. He accepted the position and held it the remainder of his life, beginning with the rank of instructor and being steadily promoted until he reached a full professorship in 1925.

Three years (1912–15) were spent in graduate work at Washington University (St. Louis) where he held the Rufus J. Lackland Fellowship in the Henry Shaw School of Botany. As an undergraduate he had developed an interest in the "pore fungi." With the aid and encouragement of such men as Fink, Murrill, Bresadola, and C. G. Lloyd he wrote and published his first paper entitled "The Known Polyporaceae of Ohio" at the end of his junior year (June, 1911). This is further evidence of his early scientific maturity. His ability for taxonomic work with the fungi and his interest in the polypores were guided and heightened by association during the graduate years with Dr. E. A. Burt, of the Missouri Botanical Garden. Three important and extensive papers on the Polyporaceae followed in 1914 and 1915 and thoroughly established him as a specialist in that group. Although he worked with all fungi and had an amazing knowledge of all groups, he retained his love for the polypores to the last. He left a voluminous manuscript for a manual of the pileate Polyporaceae of North America north of Mexico practically ready for publication. This represents a compelling interest over a 35-year period of activity.

His Ph.D. degree was conferred by Washington University in 1915. Coming to Penn State that year he first taught general botany and later took over the courses in mycology and tree diseases. He was an excellent teacher. In 1936 he was a joint-author of the HILL, OVERHOLTS and POPP, *Botany, a textbook for colleges*. His knowledge of the fungi especially fitted him for development of the courses in forest pathology. He prepared his own manual for that work and brought together abundant illustrative materials. His graduate courses were always popular and his students came to share his enthusiasm.

Overholts was a great collector. He had a passion for field work and a remarkable ability for inspiring those who accompanied him. He knew where to go not only in the vicinity where he lived but was familiar with many distant localities. Many members of the Mycological Society of America know how much he contributed to the success of the forays which he attended. He was a great believer in photographic records of his fungi. He did his own photography and it could not have been done better. The carefully indexed file of photographs which he made is amazing both for its quality and its magnitude. Looking over the photographic file and then turning to tome after tome of notes and drawings one wonders how time was found for such comprehensive achievements. In addition there were cultures which demanded much time and attention for preparation, care and study. However there was time for all these achievements and also time for recreation for until his health failed him he was an ardent trout fisherman and he always looked forward to the opening of the hunting season.

In the Mycological Society of America, Dr. Overholts served as vice president in 1937, president in 1938, and councilor in 1939. He was a member of Sigma Xi; the Pennsylvania Academy of Science; the Torrey Botanical Club; Phi Beta Kappa; Alpha Zeta; Phi Eta Sigma; Gamma Sigma Pi; and Phi Kappa Tau.

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TAXONOMIC NOTES ON LOUISIANA FUNGI—I

LINDSAY S. OLIVE

(WITH 3 FIGURES)

This paper is one of a proposed series of papers in which the writer plans to present new data on the fungus flora of Louisiana. The description of new species as well as the report of new host records and noteworthy information on distribution of the fungi will be included. Both saprophytic and parasitic species will be considered. The present paper includes a description of several new genera and species of parasitic fungi found in Louisiana, in addition to new distributional and host records for several other species.

PLASMOPARA GONOLOBI (Lagerh.) Swingle

This downy mildew was collected on a species of *Gonolobus* (*Vincetoxicum*). Most of the diseased leaves were also rather heavily covered with the dark brown telia of *Puccinia obliqua* B. & C. The mildew appeared in conspicuous white fuzzy coalescing patches on the under surface of the leaves. Considerable defoliation resulted.

Collected on *Gonolobus* sp.; Baton Rouge, Louisiana; October 23, 1946.

PERONOSPORA PARASITICA (Pers.) Tul.

This mildew, which agrees well morphologically with the descriptions of *P. parasitica,* was found parasitizing an unreported host, *Cleome spinosa* L. This seems to be the first report of the mildew on a member of the Capparidaceae, a family related to the Cruciferae. A downy growth of conidiophores was observed on the under side of the lower leaves. The sporangia (conidia) measured 16–20.4 × 20.4–27 μ. Iwata (1937) refers to a *Peronospora*

sp. on *Cleome spinosa,* but we have no other information on it than this. He apparently did not identify the species of mildew.

Collected on leaves of *Cleome spinosa* L.; Shreveport, Louisiana; December 23, 1946; W. J. Dickson.

PERONOSPORA POTENTILLAE deBary (FIG. 3, *A*)

This mildew was found on the leaves of dewberry, almost entirely on those leaves already attacked by a rust, *Kunkelia nitens.* The mildew appears as a fuzzy grayish or slightly violaceous growth on the leaves, where it generally produces irregular brown spots of various sizes. Sometimes entire leaves or young shoots are killed by the rust-mildew combination.

The sporangiophores are 5–7 times dichotomously branched, 7.8–11.3 μ in diameter at the base, 535–695 μ in overall height, their stalks 340–575 μ long, and the ultimate branches pointed, recurved, and 5–16.5 μ long. The sporangia (conidia) are oval to ellipsoidal, with pale sordid-violaceous walls, measure 14–18 × 19–29 μ, and germinate with germ tubes.

Wilson (**1914**) recognized three species of *Peronospora* on rosaceous hosts in America. These species were listed as *P. potentillae* deBary, *P. rubi* Rabh., and *P. fragariae* Roze & Cornu. He considered *P. rubi* to be the only species of mildew on *Rubus* in this country. Davis (**1914, 1919, 1924**) reported *P. rubi* on several species of *Rubus* in Wisconsin. The mildew seems to be the same as ours. Fischer (**1892**) considered all three of the above mentioned species of *Peronospora* synonymous and included them all under the name *P. potentillae,* this name antedating the others. According to him, the only other valid species on members of the Rosaceae is *P. sparsa.*

I am more inclined to agree with Fischer in considering *P. rubi* synonymous with *P. potentillae.* Our fungus actually fits better into deBary's original description of the latter than it does into Rabenhorst's description of *P. rubi.* Unless experimental results prove otherwise, I believe that all these collections of downy mildew on *Rubus* should be classified as *P. potentillae* deBary, of which *P. rubi* is probably best considered a synonym.

Parasitic on leaves of *Rubus* (probably *R. trivialis*); Baton Rouge, Louisiana; April 11, 1947.

MICROSPHAERA ALNI var. **cinnamomi** var. nov. (FIG. 1: 11–15)

Peritheciis amphigenis, disseminatis, 77–142 μ in diametro; parietibus cellularum 10.4–21 μ in diametro, ascos 2–8 continentibus; appendicibus 1.5–2 × diametrum peritheciis, 137–306 μ longis, 1–6 × dichotome ramosis (raro haud ramosis), apicibus ultimis non recurvis. Ascis 27.8–46.1 × 47–67 μ, 2–7 sporogenis; ascosporis 10.5–16.5 × 18.3–25.5 μ. Parasitica in plantulis juvenilibus *Camphorae cinnamomi* Nees & Eberm.

Perithecia scattered over upper and lower surfaces of the leaves, 77–142 μ in diameter, wall cells 10.4–21 μ in diameter, containing 2–8 asci; appendages 1.5–2 times the diameter of the perithecia, measuring 137–306 μ in length, 1–6 times dichotomously branched, rarely unbranched, the ultimate tips not recurved. Asci short-stalked, measuring 27.8–46.1 × 47–67 μ, 2–7 spored; ascospores 10.5–16.5 × 18.3–25.5 μ.

Parasitizing young seedlings of *Camphora cinnamomi* Nees & Eberm.; Baton Rouge, Louisiana; February, 1947; Q. S. Holdeman.

The diseased seedlings were found growing beneath the parent tree. The mildew produced grayish white spots on both leaf surfaces, or often covered almost the entire leaf surface. Conidia were not being produced, but a few old conidia, typical in form, were observed under the microscope.

Salmon (**1900**), in his monograph of the Erysiphaceae, lists several varieties of *M. alni* and points out that the species is probably the most variable in the Erysiphaceae. Apparently, the most important single consideration in distinguishing the different species and varieties of *Microsphaera* is the character of the appendages, and even the appendages often show considerable variation within the species or variety.

The present fungus differs from typical *M. alni* and its known varieties in the more variable mode of branching of its appendages, in the failure of the ultimate branches to show any recurving, and in its parasitism of camphor seedlings. The present variety approaches the description of *M. alni* var. *lonicerae* (DC.) Salm., the appendages of which generally show no recurving of the ter-

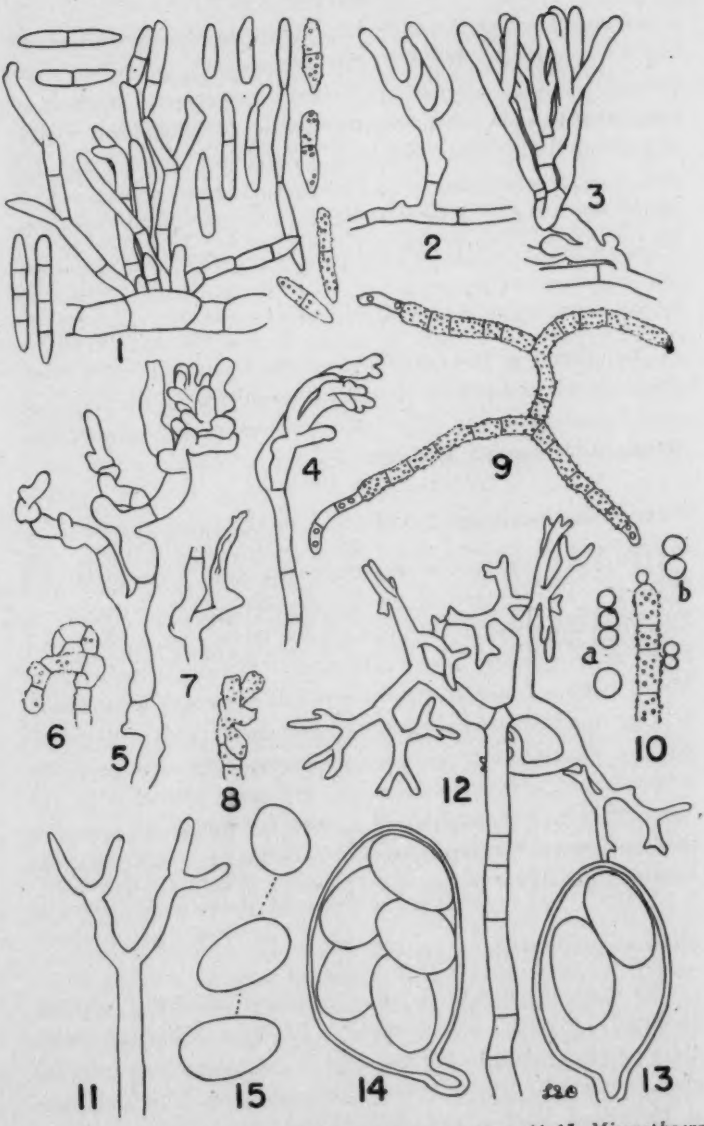

FIG. 1. 1, *Ramularia sidae;* 2–10, *Thallospora aspera;* 11–15, *Microsphaera alni* var. *cinnamomi.*

minal branches, but the latter is parasitic on species of *Lonicera* and has appendages only 3–4 times dichotomously branched. The two differ in a few other details of minor importance. It would be interesting to know whether these varieties of *M. alni* are capable of growing on the hosts of typical *M. alni* such as alder and oak and, if so, whether they would continue to exhibit what we are considering varietal differences, or whether they would revert to the typical form. Experimental evidence is needed here.

BUBAKIA CROTONIS (Cke.) Arth.

This interesting rust was found on the leaves of *Croton punctatus*. Uredinia and telia occurred on both sides of the leaves on small brown leaf spots. The collection was made at Grand Isle, Louisiana; November 30, 1946.

PHYSOPELLA FICI (Cast.) Arth.

This rust is common in our area on leaves of *Ficus carica* and appears to be the cause of considerable defoliation of that plant. The uredinial phase is abundant on the leaves, but we have not found telia. We also have a collection of this rust (uredinia) on *Morus rubra*. Although there are earlier reports of it on *Morus* in other countries, we have found no previous records of its occurrence on this particular species of mulberry. The uredinia in this collection are brown, amphigenous, but more abundant on the lower surface, with paraphyses around the periphery, and with uredospores that measure 14.4–22.5 × 20.6–34 μ. The collections were made at Baton Rouge, during the fall months of 1946.

USTILAGO SPHAEROGENA Burrill

This smut, affecting the heads of a grass, *Echinochloa colonum*, produced spores that measured 6–7 × 7.1–8.7 μ. Diseased grains are somewhat enlarged, but the smut is not conspicuous until the grain is crushed. The collection was made by Dr. Clair A. Brown at Thibodaux, Louisiana; October, 1946.

Ramularia sidae sp. nov. (FIG. 1:1)

Hypophyllis, areolas pulveraceas et albas proferentibus, 1–5 mm. in diametro; conidiophoris racematis per stomata perrumpentibus, simplicibus vel ramosis, septatis; conidiis a termino latereque gestis, saepe catenulatis, cellulas 1–4 habentibus, 2.3–4.6 × 10.4–40 μ, maturis parietis asperos frequenter gerentibus. Parasitica in foliis *Sidae* sp.

Forming powdery white patches on the under surface of the leaves, measuring 1–5 mm. in diameter; leaf tissue generally not discolored, or only slightly yellowish in spots where the fungus is sporulating; symptoms generally not apparent on the upper leaf surface. Conidiophores arising in clusters through the stomates, simple or branched, septate, producing conidia terminally and laterally, conidia frequently catenulate, 1–4-celled, measuring 2.3–4.6 × 10.4–40 μ, often with small scattered warts over the surface at maturity.

On leaves of *Sida* sp.; Baton Rouge, Louisiana; January 13, 1947.

This seems to be the only species of *Ramularia* described on *Sida*, and it is apparently a distinct new species. A specimen of *Ramularia* on an unidentified species of *Sida*, collected by Shear at Winter Park, Florida, was recently forwarded to me by Mr. John A. Stevenson. The fungus proved to be identical with ours.

Thallospora gen. nov.

Parasitus systemicus, sine conidiophoris bene delineatis, conidiis ex hyphis ramosis excrescentibus, gracilibus, dichotome ramosis, multiseptatis, in massa alba intra hospitis ovarium factis.

Systemic parasite; no well-developed conidiophores present, conidia developing as direct outgrowths from branching hyphae, slender, dichotomously branched, multiseptate, hyaline, produced in a white mass inside the ovary of the host.

The type species is *T. aspera*.

The genus is placed in the family Moniliaceae of the Moniliales. There has been some doubt as to what tribe of that family would best accommodate the genus. The Staurosporae, as defined by Clements and Shear, would appear to be the most satisfactory. According to these authors, the tribe is characterized by "conidia forked or lobed, radiate or stellate, hyaline or bright colored, septate or continuous." The conidia of our new fungus are usually

several times dichotomously forked, the branches typically spreading out in radiate fashion.

Thallospora aspera sp. nov. (FIGS. 1:2–10; 3, *B–E*)

Parasitus systemicus et obligatus. Hyphae per maximum partem hospitis penetrantibus, in ovario sporogenis; conidiis in massa alba in ovario effectis, gracilibus, dichotome ramosis, multiseptatis, parietes asperos habentibus, paulum hygroscopicibus, 68–237 μ longis, ramis 4.5–9.5 μ in diametro; germinantibus ut tubulos germinum breves et septatos efficiant, vel microconidia globosa ex ulla cellula gerentibus, microconidiis 2.5–5.2 (6.5) μ in diametro. Parisitica in *Veronica peregrina* L.

A systemic and obligate parasite, with hyphae penetrating throughout most of the host, sporulating in the ovaries of the host; conidia produced as direct outgrowths of branching hyphae, breaking away at maturity and filling the capsule, several times dichotomously branched, multiseptate, rough-walled, somewhat hygroscopic, measuring 68–237 μ in length, the branches 4.5–9.5 μ in diameter, germinating to produce short septate germ tubes from the ends of the branches, or budding out small globose conidia terminally and laterally, microconidia measuring 2.5–5.2 (6.5) μ in diameter.

Parasitizing *Veronica peregrina* L.; Baton Rouge, Louisiana; March–April, 1947.

The fungus was just called to the writer's attention by Dr. C. W. Edgerton, who had observed it for a number of years around Baton Rouge. Infected plants in the field are not distinguishable from diseased plants until the capsules open. Then it can be seen that the capsules of infected plants are filled with white masses of conidia in which tiny brown, aborted seeds are imbedded. Generally all capsules of an infected plant become filled with conidia. Free-hand sections through various parts of these plants show that the hyphae may be present anywhere except in the smaller roots. They appear in the upper part of the primary root, in the stem up to the top of the plant, frequently in the leaves, and in all parts of the flower. Hyphae pass from the stem through the peduncle of the flower and then into the petals, ovary, style, stigma, and through the filaments of the stamens into the anthers. In the anthers, some of the hyphae wind about among the pollen grains and apparently destroy a number of them.

The organism sporulates only in the ovary. Here the hyphae penetrate mainly through the septum and placentae out into locules. Some enter the funiculi of the ovules. These developmental phases may all be observed in young flower buds several days before they open. Up to this stage of development all hyphae have been intercellular. No haustoria are produced by the fungus. Soon after hyphae penetrate the funiculus, a few of them continue on up into the tissue of the ovule itself, mostly in the nucellar region at first. Later, when the seeds are approaching maturity, the hyphae within them are observed to be much more abundant. Some penetrate the endosperm, while others may enter the embryo. At this time, some of the hyphae within the seed are seen to be intracellular, narrowing considerably where they pass through the cell walls. By the time the capsule opens, the seeds all appear brown, hard. and somewhat shrunken. None of them seems to be capable of germinating. However, it is possible that these aborted seeds, which fall to the ground, may serve as a means by which the fungus is carried over in the soil until the new infections take place.

During these developments within the ovules and seeds, the hyphae which have passed out into the locules repeatedly fork in dichotomous fashion to produce fascicles of branches (FIG. 1: 2–4). At maturity the fascicles break up into their component parts, the conidia. The conidia are hyaline, rough-walled, multiseptate, and dichotomously branched. According to Vuillemin's definition, such conidia, which are actually a part of the hyphae bearing them and not distinct structures formed *de novo,* are termed *thallospores.* Remains of the hyphae which give rise to the conidia may be found in the ovary after the conidia are shed (FIG. 1: 5–8).

When placed in water the conidia show a hygroscopic tendency to spread out in radiate fashion (FIG. 1: *C, D*). On nutrient agar, they may germinate terminally to produce short septate germ tubes, which cease developing early (FIG. 1: 9). In water and on some agars numerous spherical microconidia are budded out from any cell (FIGS. 1: 10; 3, *E*). They measure 2.5–5.2 (6.5) μ in diameter. On agar some of the cells of the macroconidia swell into chlamydospore like structures (FIG. 3: *E*). All attempts to grow the fungus in culture have failed.

I have studied some slides of the fungus prepared by Dr. M. T. Cook with the iron alum haematoxylin technique. Sections through diseased ovaries of the *Veronica* revealed the presence of one to several nuclei (mostly 2–4) in each hyphal cell, and one or two nuclei in each cell of the macroconidia.

Greene (**1943**) reports finding this same fungus on *Veronica peregrina* in Wisconsin, but he makes no attempt to classify or formally describe it. He thought that the fungus occurred only in the fruits and seeds. He was unable to find hyphae passing through the flower peduncles.

Ormathodium ambrosiae sp. nov. (FIG. 2: 1–11)

Maculis hypophyllis; 1–5 mm. in diametro; fungo in forma incrementi plumosi, purpureobrunnei se ostendenti; hyphis in folio numerosis, inter cellulas sitis; conidiophoris per stomata emergentibus, subbrunneis, aliquando roseis, multiseptatis, semi-erectis, super pilos folii saepe ascendentibus atque ab apicibus eiusdem se diffundentibus. Conidiis a termino latereque singulis aut duobus tribusve catenatis, in colorem subbrunneum se vertentibus, 1–5 septatis, 3.8–7.3 × 22–68 μ, a termino vel latere germinantibus. Parasitica in foliis *Ambrosiae trifidae* L.

Spots chiefly hypophyllous, 1–5 mm. in diameter, fungus appearing as a fuzzy purplish brown growth; hyphae abundant in the leaf, intercellular; conidiophores emerging through the stomates, chiefly on the under surface of the leaf, brownish, sometimes rose-tinted, multiseptate, semi-erect, often climbing up the leaf hairs and spreading out from their tips. Conidia borne terminally and laterally, singly or in chains of 2 or 3, becoming brownish, 1–5-septate, 3.8–7.3 × 22–68 μ, germinating terminally or laterally.

Parasitic on leaves of *Ambrosia trifida* L.; Baton Rouge, Louisiana; September–October, 1946.

The fungus was obtained in culture on nutrient agar, but grew very slowly in the form of whitish or almost cream-colored yeast-like colonies. The colonies were composed chiefly of slender filiform conidia produced in great abundance on the hyphae. Inoculations were not attempted.

Sydow (**1928**) founded the genus *Ormathodium* to accommodate a parasite, *O. styracis*, on *Styrax argenteum*. It seems to be similar in several ways to our fungus on ragweed. According to Sydow, *O. styracis* has olive brown, 1–2-septate conidia in simple

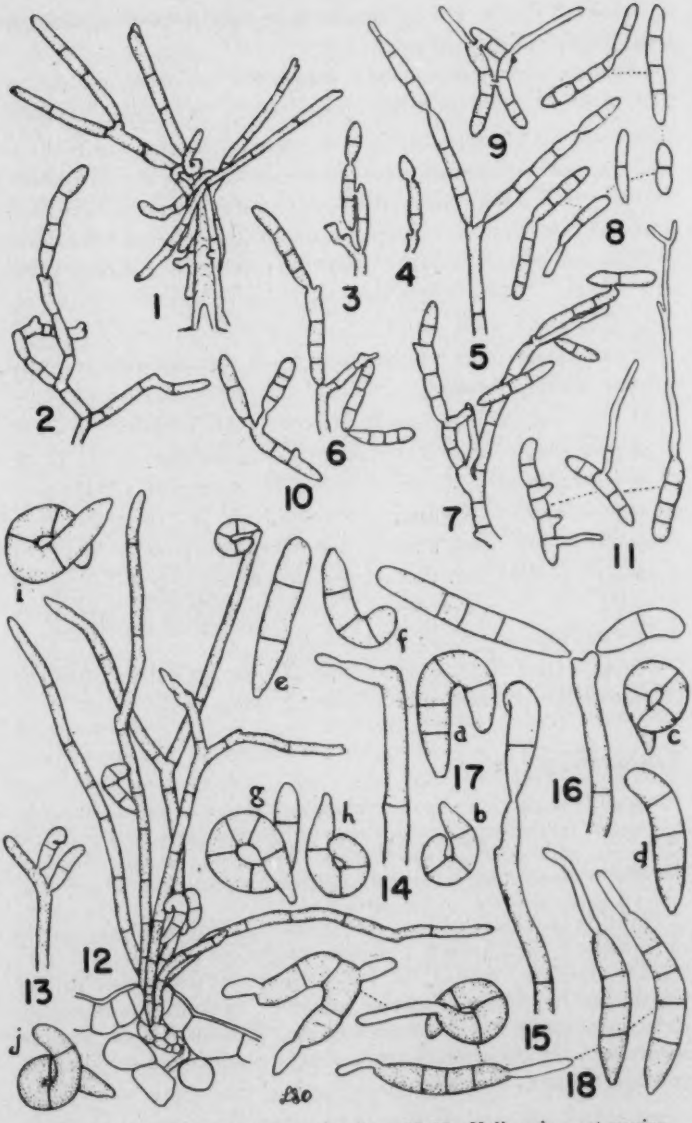

FIG. 2. 1–11, *Ormathodium ambrosiae;* 12–18, *Helicomina caperoniae.*

or branched chains, and its conidiophores also tend to spread out from the tips of the leaf hairs.

Clements and Shear (**1931**) reduce the genus *Ormathodium* Syd. to synonymy with *Dendryphium* Wallr. However, *Dendryphium* was founded on a saprophytic species (*D. comosum* Wallr.), and practically all species placed in the genus since then have been saprophytes. Even some of these saprophytes appear to have been incorrectly put into this genus. *Ormathodium* differs from *Dendryphium* in at least one very important feature, its distinctly parasitic nature. This would appear sufficient reason for considering *Ormathodium* a valid genus. To place parasitic forms in *Dendryphium* would serve only to increase the confusion already existing within that genus.

At least one other previously described species placed in *Dendryphium* should be transferred to *Ormathodium*. This is *D. costaricense* Spegaz. It was described by Spegazzini (**1919**) as a parasite on leaves of *Xylosma Salzmanni* Eichl. in Central America. Saccardo (**1931**) has already questioned its position in *Dendryphium*, pointing out that its parasitic nature might distinguish it as the type for a new genus. Saccardo appears to have overlooked Sydow's genus, *Ormathodium*.

I propose the following new combination for Spegazzini's fungus: **Ormathodium costaricense** (Spegaz.) comb. nov.

Helicomina gen. nov.

Parasiticis in plantis generosis; conidiophoris elongatis, fuscis, simplicibus vel ramosis, multiseptatis; conidiis a termino latereque partis, rectis, curvatis, glomeratis, pro longitudine validis, haud hydroscopicis, septatis, coloratis. Coloniae in formis fuscarum areolarum plumosarum praecipue se ostendunt.

Parasitic on higher plants; conidiophores elongate, dark, simple or branched, multiseptate; conidia produced laterally and terminally, straight, curved, or coiled, stout in proportion to the length, not hygroscopic, septate, colored. Colonies appearing as dark fuzzy patches or sometimes effuse. Except for its parasitic nature and the presence of straight as well as coiled conidia, the genus resembles *Helicoma*.

The type species is *H. caperoniae*.

Helicomina caperoniae sp. nov. (FIGS. 2: 12–18; 3: *F, G*)

Praecipue hypophyllis, maculas fuliginosas, 4–10 μ in diametro, efficientibus vel effusis; mycelio inter cellulas sito; conidiophoris per stomata racematis, simplicibus vel subramosis, brunneis, 2–12-septatis, 4–6.6 × 92–329 μ, conidia et a termino et a latere ferentibus; conidiis rectis, curvis, vel glomeratis, 1–7-septatis, plerumque triseptatis, pallido-brunneis, hilum parvum habentibus, 4.5–6.3 × 17.4–40 μ, spiris 12.2–19.1 μ in diametro, terminibus vel latere germinantibus. Parasitica in foliis *Caperoniae castaneaefoliae* (L.) St. Hil.

Chiefly hypophyllous, producing sooty spots 4–10 mm. in diameter, or becoming effuse; internal mycelium intercellular, without haustoria; conidiophores appearing in clusters of several through the stomates, simple or few-branched, brown, mostly 2–12-septate, measuring 4–6.6 × 92–329 μ, bearing the conidia both terminally and laterally; conidia straight, curved, or up to 1⅓ times coiled, 1–7-septate, mostly 3-septate, pallid brown, with a small hilum at the proximal end, the straight conidia measuring 4.5–6.3 × 17.4–40 μ, the coils (measured at right angles to the long axis of the conidiophore) 12.2–19.1 μ in diameter, germinating at both ends and sometimes laterally.

Parasitic on leaves of *Caperonia castaneaefolia* (L.) St. Hil.; Baton Rouge, Louisiana; October 2, 1946; Q. L. Holdeman.

The genus *Helicomina*, as defined here, is distinguished from *Helicoma* chiefly on the basis of its distinctly parasitic nature. The genus *Helicoma*, as defined by Linder (**1929**), is made up of saprophytic species. Our fungus further differs from all known species of *Helicoma* in its tendency to produce a large number of straight conidia in addition to the curved or coiled ones. This latter character, however, should probably not be considered of much generic value.

In his key to the genera of the Helicosporous Fungi Imperfecti (**1929**) and in a later paper (**1931**), Linder distinguishes the genus *Helicoceras* (*Gyroceras*) from the other genera mainly on the basis of its parasitism of vascular plants and in its possession of toruloid spores and reduced conidiophores. Our fungus differs from *Helicoceras* in the possession of well-developed conidiophores and in the failure of the conidia to show any conspicuous constrictions in the region of the septa. Although the conidia of *Helicomina caperoniae* produce short germ tubes when they are placed in water or on nutrient media, they cease developing early. Attempts to grow the fungus in artificial culture have failed.

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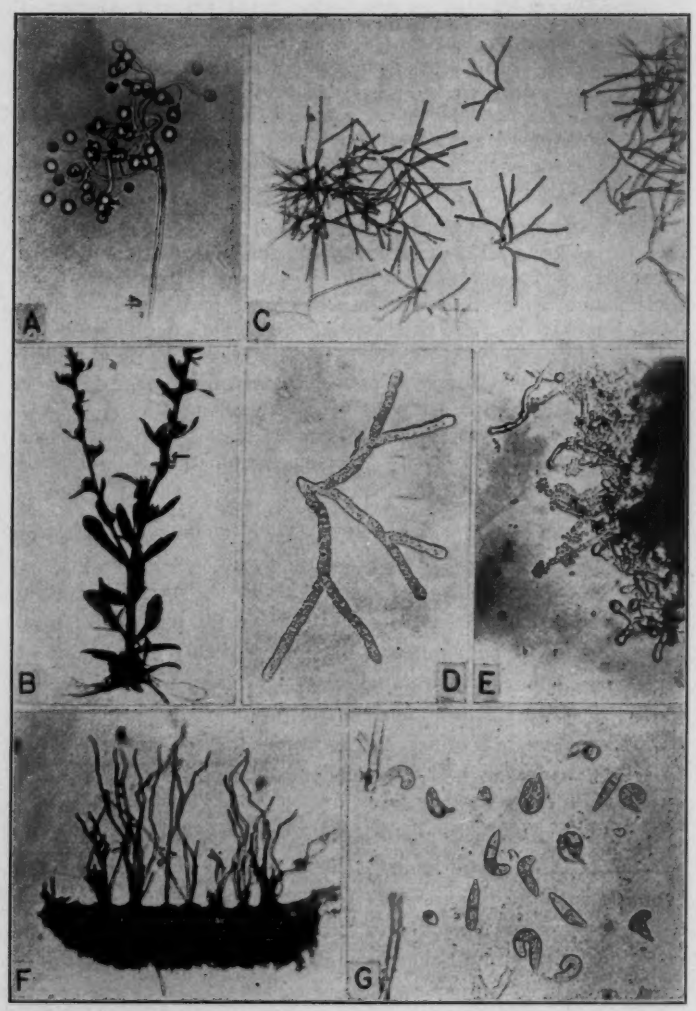

FIG. 3. A, *Peronospora potentillae;* B–E, *Thallospora aspera;* F, G, *Helicomina caperoniae.*

The writer is grateful to Dr. C. W. Edgerton for making the photographs, to Dr. P. G. Moorhead of this university for preparation of the Latin descriptions, and to Mr. John A. Stevenson of the Beltsville Plant Industry Station for information on host records.

Type specimens of new species of fungi described in this paper have been deposited in the herbarium at Louisiana State University and in the mycological herbarium at the Plant Industry Station of the United States Department of Agriculture, Beltsville, Maryland.

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EXPLANATION OF FIGURES

FIG. 1. *Ramularia sidae*, 1. Conidiophores and conidia. *Thallospora aspera* (2–10). 2–4, dichotomously branched hyphae developing into conidia; 5–8, hyphae after conidia have broken away; 9, conidium germinating; 10, portion of a conidial branch producing microconidia (*a*, *b*). *Microsphaera alni* var. *cinnamomi* (11–15). 11, 12, perithecial appendages; 13, 14, asci; 15, ascospores. (All drawings × 550, except 9, × 355, and 10, × 830.)

FIG. 2. *Ormathodium ambrosiae* (1–11). 1, Conidiophores diverging from the tip of a leaf hair; 2, branched conidiophore with conidium; 3, conidia fusing laterally while on conidiophore; 4, young conidium being produced apically from an older one; 5, 6, conidiophores with conidia in chains; 7, conidiophore with conidia developing at various points; 8–11, conidia, some showing anastomoses and some germinating. *Helicomina caperoniae* (12–18). 12, tuft of conidiophores arising through stoma of leaf; 13, conidiophore producing two conidia laterally; 14–16, conidiophores producing conidia terminally; 17, conidia of various shapes (*a–j*); 18, conidia germinating. (Nos. 1–13, × 360; nos. 14–18, × 650.)

FIG. 3. *Peronospora potentillae*. *A*, sporangiophore. *Thallospora aspera* (*B–E*). *B*, diseased plant, showing white mass of conidia in the seed capsules (arrow); *C*, group of mature conidia; *D*, single enlarged conidium; *E*, conidia on agar, showing the production of microconidia and chlamydospore-like swellings. *Helicomina caperoniae* (*F*, *G*). *F*, transverse section of leaf showing clusters of conidiophores arising through the stomates; *G*, conidia.

TELIOSPORE DISCHARGE IN PUCCINIA TUMIDIPES PECK [1]

STUART M. PADY [2]

(WITH 19 FIGURES)

In the Uredinales the discharge of teliospores has not been reported as far as the writer has been able to determine. The occurrence of discharged teliospores of *Puccinia tumidipes* Peck on the leaves of *Lycium halimifolium* (Matrimony Vine) in the vicinity of the telial sorus is therefore of interest.

Puccinia tumidipes is known only in the uredial and telial stages although Arthur [3] has described the pycnial and aecial stages on the basis of a single culture made from teliospores on *L. pallidum* sown on *L. halimifolium*.[4] From two sowings, a few pycnia were produced in each case and in one a single uredial sorus was formed. The rust was therefore considered to be autoecious although no field collections of the aecial stage have ever been made. On the campus of Kansas State College there is a large clump of the host which has been there for many years and which is rusted year after year. Uredia and telia appear early in the growing season and the heavily infected leaves tend to become yellow and to be cast prematurely, particularly when the weather turns hot and dry. Usually there is a second period of infection late in the growing season. Other bushes on the campus and in the town are free from rust suggesting that in this long-established clump of bushes the rust is able to overwinter, presumably in the uredial stage.

[1] Contribution No. 488, Department of Botany and Plant Pathology, Kansas State College, Manhattan, Kansas.

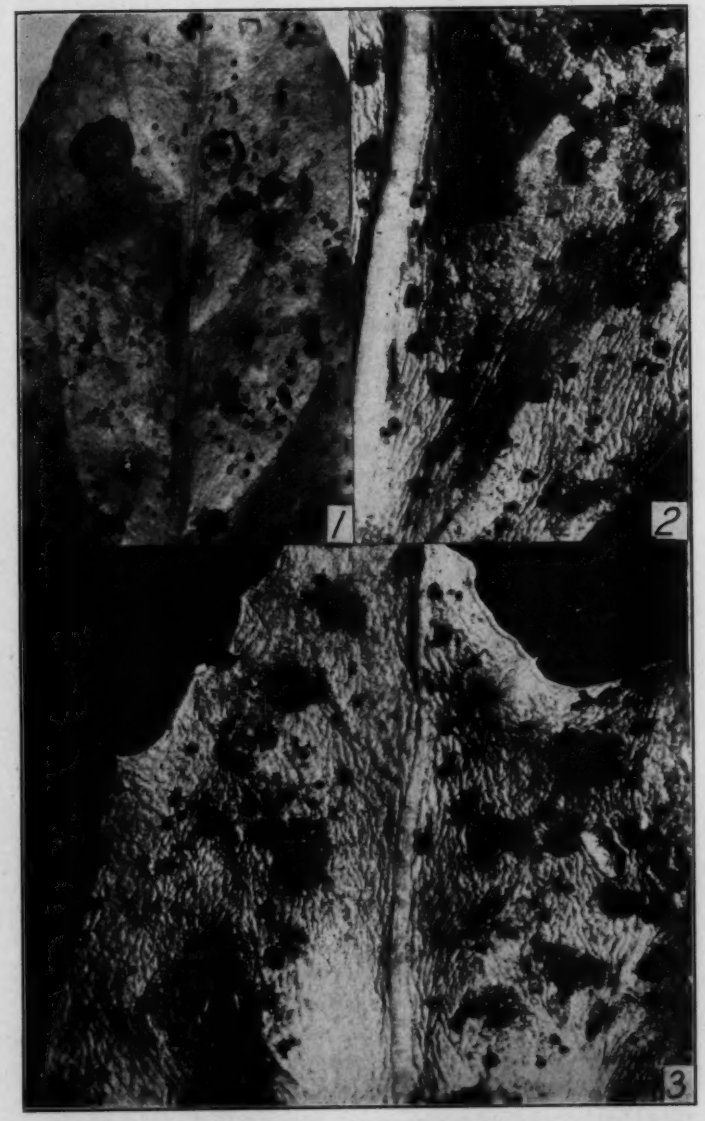
[2] This work was done while the author was Assistant Professor in the Department of Botany and Plant Pathology, Kansas State College.

[3] Arthur, J. C. Manual of the rusts of the United States and Canada. Purdue Research Foundation, 1934.

[4] Arthur, J. C. Cultures of the Uredineae in 1915. Mycologia **8**: 136. 1916.

On June 14, 1946, primary and secondary uredia were abundant on the lower surface of many of the leaves, with here and there an occasional black telial sorus (FIG. 1). Occasionally the telia were mixed with the uredia but usually they were separate. In the vicinity of some of the telia an unusual darkening of the surface was noted. The discoloration was on one side of the sorus extending fan-wise 1 to 5 mm. (FIGS. 1–3). It was black, granular, fairly uniform and had the mold-like appearance of one of the Dematiaceae of the Moniliales although the leaf tissues below were green and apparently normal. When portions were removed for examination it was discovered that it was firm and crust-like, even semi-elastic, and the removal of a small portion was likely to dislodge the remainder. The constituent particles were so firmly fused to one another that it suggested the drying down of a gelatinous mass. Under the hand lens small black shining bodies could be seen, which under the microscope were shown to be teliospores. Since this suggested that the teliospores had been discharged from the surface a careful examination of the discharged spores was made in an attempt to determine the mechanism involved.

The most striking feature of the discharged teliospores was that some had pedicels whereas others apparently lacked this structure: yet even in these there were indications of the remains of the pedicel in the form of a transparent membranous material surrounding the spore (FIG. 9). This membrane was attached at the basal end of the teliospore but free at the upper end (FIG. 17*C*), suggesting that the pedicel had actually reversed itself around the spore. This unexpected activity of the pedicel was confirmed by later observations and experiments. The discharged spores were therefore of two types, those with pedicels in normal position, and those whose pedicels surrounded the spore: for convenience the former will be referred to as pedicellate, the latter as reversed. Moreover, the pedicellate spores invariably possessed ruptured pedicels which had been broken near the base. A normal teliospore when removed from a sorus has a pedicel longer than the spore with an upper expanded portion occupying about two-thirds or more of the length and a lower unexpanded portion which is narrow, tapering, and has a roughened surface (FIG. 12). Discharged



FIGS. 1–3. *Puccinia tumidipes.*

spores usually have the pedicel ruptured at the junction of these two regions (FIGS. 13, 14).

Valuable information was obtained by mounting the discharged spores from the dry film on a dry slide, adding a cover-slip and making drawings and measurements *before* water was added. When water was run under the cover-slip two remarkable activities took place: first, the spores in the film moved rapidly apart and were often pushed completely out of the field and secondly, the spores with pedicels snapped downward and the pedicel suddenly turned inside out. In several cases it was possible to make camera lucida drawings of individual dry pedicellate spores (FIGS. 17*A*, 18*A*), then drawings of the same spores after the pedicels had reversed (FIGS. 17*B*, 18*B*). From many such observations it was concluded that the structure of the pedicel accounted for both of these activities. There is an outer, thin, tough, elastic layer, and an inner layer of highly hygroscopic material which is indicated in the drawings by a dotted line (FIGS. 12–19). When a spore is discharged the pedicel ruptures at the base and under certain conditions turns inside out surrounding the spore. On the leaf both types are found (FIG. 9) but the reversed type usually predominates.

This inner hygroscopic layer can be demonstrated by mounting the pedicellate spores in lactophenol with a trace of cotton blue and warming the slide to drive off the water and stain the spore. When this is done this layer is clearly shown as a spindle-shaped band extending from the top of the pedicel to the ruptured base, becoming progressively narrow toward each end (FIGS. 13, 14).

The existence of this highly hygroscopic layer was also shown by adding water to the dry discharged spores and then replacing the water by India ink (FIG. 10). By this method it was shown that this inner layer enlarges greatly where it is in free contact with the water. In pedicellate spores the exposed area is relatively small, but even here some swelling takes place (FIG. 16). In the reversed spores this inner layer is now external and water is rapidly absorbed so that it swells to many times its thickness (FIGS. 18*B*, 19). The photomicrograph in figure 10 shows this very clearly. Actual measurements show that the total diameter of the spore and two layers may be 60–65 μ with the individual

layers 15–20 μ with a maximum of 30 μ. In pure water (FIG. 17*C*) or lactophenol (FIG. 9) this layer would not be detected as it is almost perfectly hyaline and gelatinous but with India ink the boundaries could be accurately determined.

The rapid swelling of this inner gelatinous layer would explain the two visible activities which occur when water is added to the dry discharged spore layer. Since most of discharged spores are of the reversed type this inner layer is now outer, exposing a large surface area capable of rapid water absorption. This rapid swelling of the closely-packed spores in the dry film forces them apart and since each spore may reach an outside diameter including the two gelatinous layers of 65 μ, it means that a tremendous expansion takes place (FIG. 10). In nature the spore mass would be free to swell equally in all directions; on the slide, however, the expansion is restricted and the pedicels enlarge rapidly forming a flat sheet. In the pedicellate spores the rapid expansion of this layer would set up a tremendous pressure inside the pedicel. The tough outer wall is capable of resisting this pressure, but the exposed torn end of the pedicel lacks the outer wall at that point. The swelling gelatinous layer can, if the break is large enough, force itself out of the free end and thus partially relieve the pressure (FIG. 16).

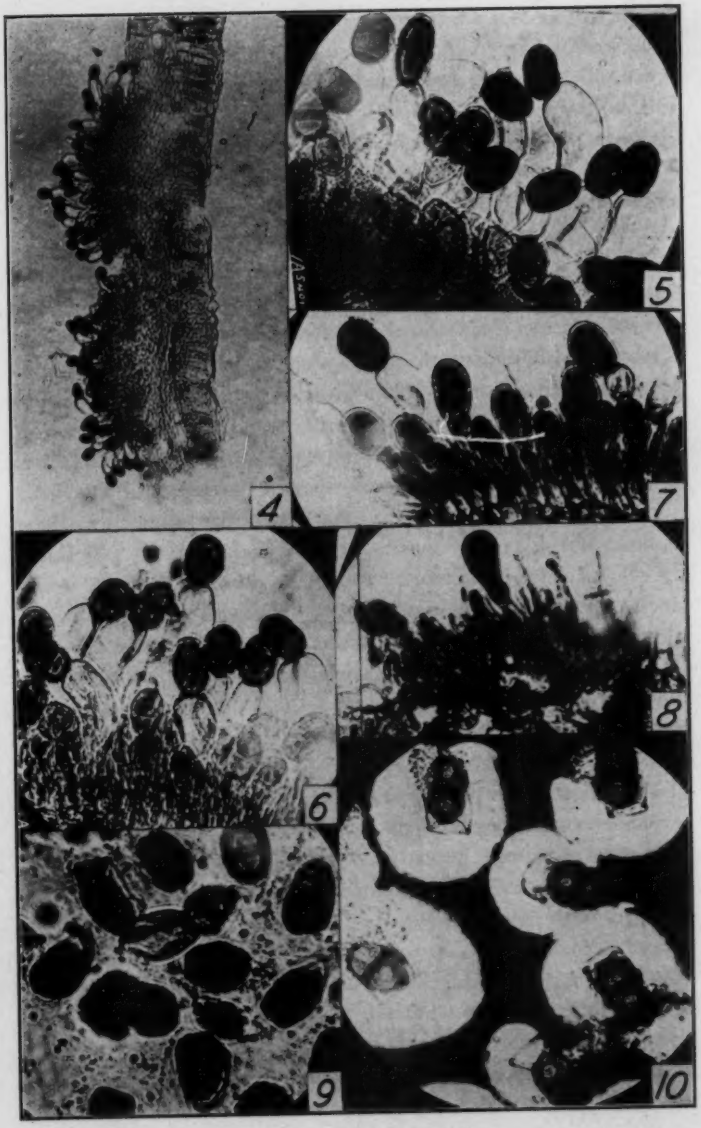
The lower exposed portion of the pedicel would develop a considerable pressure immediately, due to rapid water absorption. The pressure would be much greater than in the upper lining of the pedicel since the upper end is further from the water. Moreover the hygroscopic layer is not continuous across the upper part of the pedicel so that at the point of attachment the pressure would be practically zero. The net result of this rapid increase in pressure is a stretching of the pedicel wall with the pressure highest at the base, gradually less toward the top and zero at the extreme upper end. The lower portion opens out in a funnel-shaped manner and the lower edge begins to turn back (FIG. 15). As soon as the inner wall pressure at the free end is greater than the pressure exerted by the stretched outer wall, the outer wall is doubled backward upon itself. The pressure of the inner wall is building up so rapidly that as soon as the outer wall begins to fold back there is a sudden movement and the pedicel completely re-

verses itself. The inner wall is now on the outside and the pedicel wall stretches over the spore with a fold at the point where the outer wall doubles backward. On the slide, the cover slip and glass slide tend to hold the gelatinous inner layer in place and as a result the teliospore itself is pulled downward.

It should be emphasized that this turning inside out of the pedicel occurs so suddenly that no intermediate stages can be observed. If under low power of the dissecting microscope a discharged pedicellate spore is mounted dry on a slide with cover slip holding the specimen in place, and water is run in under the cover slip, a sudden movement of the spore takes place as soon as the water strikes the spore, and the pedicel disappears. So sudden is this transition that one can hardly believe that it has actually taken place; repeated observations however always give this almost instantaneous reaction.

Sometimes the spore turns inside out with the outer wall forming a very prominent bell where it folds backward (FIGS. 10, 18*B*). The teliospore in figure 17*A* reversed itself when water was added and a prominent bell or funnel-shaped structure appeared (FIG. 17*B*). Then almost as soon as the camera lucida sketch had been made a further shift took place and the teliospore was pulled further down inside the tube formed by the reversed pedicel wall (FIG. 17*C*).

The base of the pedicel sometimes shows an irregular surface of fine lines or ridges suggesting granules of various sizes. This is observable in dry mounts (FIG. 18*A*), in mounts in water (FIGS. 10, 15, 16), and particularly in the reversed spores (FIG. 18*B*). In lactophenol this roughened surface was not evident. The presence of this layer is of diagnostic value in determining whether or not the pedicel actually surrounds the spore, or, as was at first thought, merely lies to one side of it. The spore in figure 18*A* was mounted dry and drawn with camera lucida, water was then added with the result that the pedicel immediately reversed. The water was replaced by India ink and the spore redrawn (FIG. 18*B*); the granulations are now located around the upper part of the teliospore (FIG. 18*B*). These markings are shown clearly in the photomicrograph in figure 10, especially in the spore at the extreme left and also in the upper left hand corner.



FIGS. 4–10. *Puccinia tumidipes.*

The presence of this gelatinous layer on the outside of the reversed discharged spores explains why the spores were held in a firm film. As the conditions for discharge required the presence of abundant moisture the spores would form an irregular gelatinous group on the moist leaf surface. As the moisture disappeared the gelatinous walls would become thinner and the spores would be drawn closer together, finally forming a thin dry crust adhering tightly to the leaf surface.

Fresh material was sectioned by the free hand method, using 95 per cent alcohol to harden the material, sectioning the pith in alcohol, placing the sections in water and mounting the sections with the aid of a dissecting microscope in lactophenol and cotton blue. Photomicrographs of these sections are shown in figures 4–8. The striking feature of the free hand sections is the enormous length of the pedicels as they occur in the sorus. The spore in the upper right hand corner of figure 5 has a pedicel that measures $130 \times 28\,\mu$. It is possible that the lactophenol may have caused some additional swelling. The general appearance of such spores has a striking similarity to the elongation of the ascus immediately preceding spore discharge.

In a young sorus (FIG. 7) various stages in the development of the pedicel may be seen; the pedicel is slender at first, enlarging slightly just below the mature spore, then gradually expanding and elongating until all the spores are raised considerably above the level of the sorus (FIG. 5). Two mature sori are shown in cross section in the photomicrograph in figure 4. The elongated, swollen, more or less curved pedicels are very conspicuous here. A portion of the upper sorus of figure 4 is shown in higher magnification in figure 6. The mature teliospore may now be discharged if the right conditions of moisture are obtained. The inner wall absorbs water slowly through the wall and through connecting hyphal cells. The resulting internal pressure presumably ruptures the outer wall and the teliospore is discharged. The break usually occurs somewhere in the lower end of the pedicel: it may be just above the basal portion or higher; it never includes the basal portion. Thus in an old sorus that has discharged most of the spores, there remains a mass of torn pedicel bases which

gives the sorus a very ragged appearance, clearly shown in the photomicrograph in figure 8.

Since the pedicel probably requires free water for successful discharge an attempt was made to determine the effect of free water upon the mature sorus. A leaf with a mature sorus was placed under the high power of a binocular dissecting microscope. At this magnification and with good illumination the black, shiny teliospores could be clearly observed. The sorus was then flooded with water. Almost immediately there were signs of activity as evidenced by slight movements of spores. These movements were mostly from side to side but soon it was evident that there was a strong upward movement as well. In a few minutes all of the spores were exhibiting a twisting motion that was fascinating to observe. The spores were thus pushed upward into the drop of water. No sudden discharge was observed under these conditions although many spores became free in the drop of water.

Leaves bearing telia were placed in Petri dishes and slides suspended a short distance (2–4 mm.) above the sori. In some cases teliospores were found on the lower surface of the slide suggesting that they had been deposited there by spore discharge. The leaves, however, are not flat and are inclined to curl so that it is possible that in some cases the leaf actually touched the slide. In one experiment four pedicellate spores were found at the end of twelve hours on one slide above a heavily infected leaf. At the end of twenty-four hours over 300 were present. One series of seven Petri dishes showed teliospores on three slides at the end of twenty-four hours. Another series of seven was left for four days. At the end of that time all of the slides had spores on their lower surface: three had many, two had four to six, whereas the remainder had but one teliospore each. In six of the seven dishes urediospores were present also on the slides, which means either that the slide accidentally touched the leaf or that the urediospores had also been discharged. Since the uredia are not known to discharge their spores it suggests accidental contact.

The lower surface of the exposed slides was not coated with an adhesive, yet the teliospores were firmly attached. The teliospore itself is apparently adhesive, sticking to any surface. In the

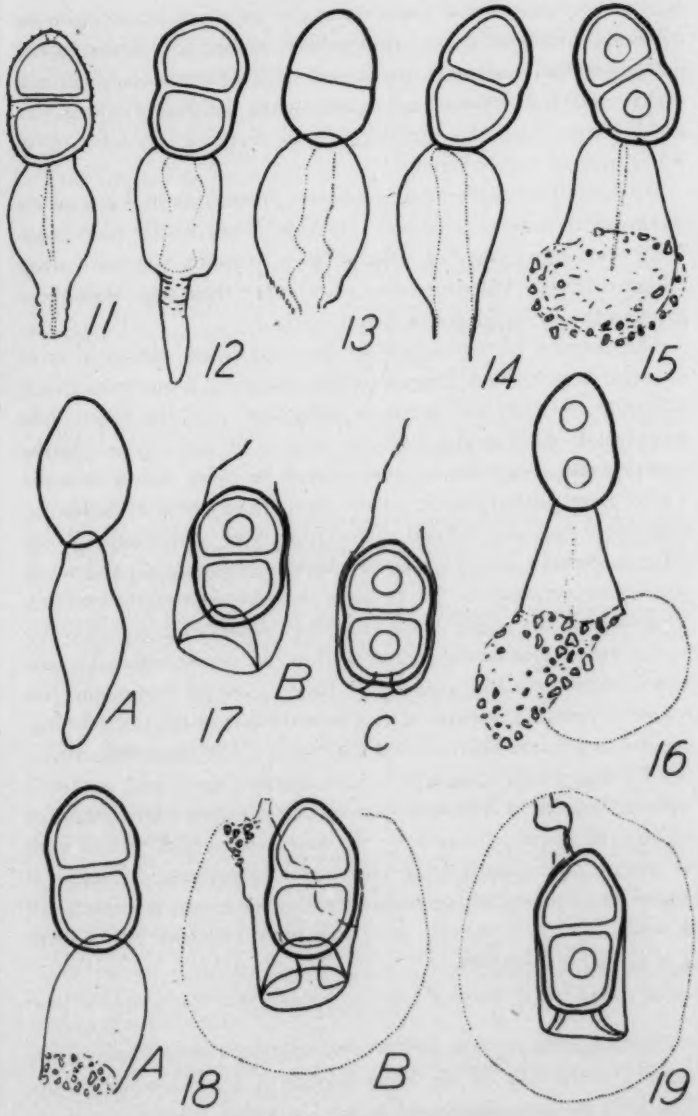
Basidiomycetes all discharged spores have been found to have sticky or moist surfaces. This would explain the adhesiveness of the discharged teliospores, but would not account for the presence of the urediospores there unless they too had a sticky surface.

DISCUSSION

The teliospore pedicel in the rusts not uncommonly swells when moistened in water. In *Phragmidium* this is the basis upon which the various sections are separated: in the Euphragmidium section "the lower half of the pedicel is hygroscopic" whereas in the section Earlea the pedicel is "firm not hygroscopic." [3] Pedicel swelling also occurs in *Gymnosporangium inconspicuum* and the pedicel in *G. claviceps* is very similar to the pedicel of *P. tumidipes*. In *Uropyxis amorphae* a very unusual situation is found in which the teliospore is hygroscopic as well as the pedicel. The teliospore has an "inner layer chocolate-brown 2.5–3 μ thick, the outer layer colorless, gelatinous, swelling in water 7–15 μ thick at sides and 1–3 μ thick at apex and base, finally bursting and disappearing; pedicel colorless, as long as the spore, deciduous or swelling in water and bursting." [3] If the pedicel bursts in water it is also probable that bursting occurs in the sorus and that discharge of the teliospores could take place. Three species of *Puccinia* are recorded [3] in which the pedicel swells in water: *P. dondiae* on *Suaeda intermedia*, *P. globosipes* [5] on *Lycium andersonii* and other species, and *P. tumidipes*. *Puccinia globosipes* is probably very closely related to *P. tumidipes*.

The discharge of teliospores is not primarily a function of the pedicel, but under certain conditions the swelling causes the pedicel to rupture, but whether this is done with considerable force is not known. In many sori loose teliospores are very numerous on the surface of the sori suggesting that pedicel rupture has merely set them free. If sufficient moisture accumulated around the sorus a film would form on the lower surface and a drop would tend to develop at that point. Teliospores would be set free in this drop, and due to the position of the leaf would spread out over the sur-

[5] *Puccinia grayiae* Arth. is listed in Arthur's Manual as having swollen pedicels. However this species is now considered to be synonymous with *P. globosipes* (G. B. Cummins: personal communication).



FIGS. 11–19. *Puccinia tumidipes.*

face to one side of the sorus. As the moisture evaporated the gelatinous mass of spores and pedicels would dry down to the flaky crust found on most dry leaves. The observations described earlier of the activity of teliospores when a drop of water was added suggest that this method of spore discharge into a drop of water may be very common.

Discharged teliospores are sometimes found in typical groups on leaves which bear no telial sori. Obviously these must have come from some other leaf but whether they came by way of a drop falling from one leaf to another or whether they were shot there through the air could not be determined.

The position of the pedicel in the discharged teliospore is of interest: in one count there were 134 spores in a group of which 120 were reversed and fourteen pedicellate. On the other hand some counts show as high as 75 per cent of the latter type. In the experiments where leaves were placed in Petri dishes and the spores were discharged on a slide the majority of the discharged teliospores had the normal unreversed type of pedicel. When these discharged teliospores are mounted dry on a slide and water added, the majority of the pedicels show an immediate reversal. This may be due simply to the extremely rapid imbibition of water by the inner layer of the pedicel wall in the region exposed to the water, namely the ruptured base. Rapid swelling ensues and tremendous pressure built up at that point results in the pedicel turning inside out and surrounding the spore. The conditions requisite for this would appear to be a discharged spore with ruptured pedicel, desiccation, followed by abundant moisture. Undischarged teliospores would be unable to do this as the intact pedicel with its narrow base effectively prevents rapid absorption of water. It would be of interest to examine other rusts whose pedicels swell in water, such as *Uropyxis amorphae,* to see whether or not there is a similar mechanism.

SUMMARY

Discharged teliospores of *Puccinia tumidipes* were found in the vicinity of the telia on the lower surface of *Lycium halimifolium*. The pedicels were discovered to have a highly hygroscopic inner layer which swells greatly in water rupturing the outer wall and

setting the spore free. Under certain conditions the pedicel reverses itself, completely surrounding the teliospore. The discharged spores are sticky, adhering to each other and to the leaf surface. This is the first report of teliospore discharge in the Uredinales.

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EXPLANATION OF FIGURES

FIGS. 1–3. Photographs of lower surface of leaves of *Lycium halimifolium* showing discharged teliospores in black, fan-shaped areas adjacent to telia.

FIG. 1 × 4 (approx.).
FIGS. 2, 3 × 15 (approx.).

FIGS. 4–8. Photomicrographs of freehand cross sections of fresh material showing mature teliospores with greatly elongated pedicels. Figure 7 is a section through a young telium; figure 8 is through an old telium and shows ruptured pedicel bases.

FIG. 9. Photomicrograph of discharged teliospores with reversed pedicels from leaf surfaces mounted in lactophenol and stained with cotton blue. Note lack of pedicel swelling in the reversed spores, and also the presence of a single pedicellate teliospore.

FIG. 10. Photomicrograph of discharged teliospores with reversed pedicel from leaf mounted in water, later replaced by India ink. Note enormous swelling of pedicel wall.

FIG. 4 reduced to × 40 (approx.).
FIGS. 5–8 reduced to × 200 (approx.).
FIGS. 9, 10 reduced to × 250 (approx.).

FIG. 11. Normal teliospore and pedicel (from Arthur's Manual).

FIG. 12. Teliospore in lactophenol with intact pedicel. Hygroscopic layer shorter than normal.

FIGS. 13, 14. Discharged teliospores mounted in lactophenol. Note ruptured base. Inner margin of hygroscopic layer shown by dotted lines.

FIG. 15. Discharged pedicellate spore mounted in water showing swollen ruptured base with granulations.

FIG. 16. As in figure 15, hygroscopic layer bulging outward.

FIG. 17. *A*, discharged spore mounted dry; *B*, same a few seconds after water added, pedicel now reversed; *C*, same a minute later. Outer margin of pedicel not visible.

FIG. 18. *A*, discharged teliospore mounted dry. *B*, same spore a few seconds after water added. India ink used to determine outer margin of pedicel wall (shown by dotted lines).

FIG. 19. Teliospore from leaf surface completely surrounded by reversed pedicel. Gelatinous layer 18 μ thick: total width 65 μ.

FIGS. 12–19 drawn by camera lucida. Magnification × 520.

ASSAY OF CELLULOLYTIC ACTIVITY OF MOLDS ISOLATED FROM FABRICS AND RELATED ITEMS EXPOSED IN THE TROPICS *

W. Lawrence White, Richard T. Darby, Gladys M. Stechert, Kathryn Sanderson

(with 3 figures)

INTRODUCTION

Cotton fabrics exposed in nature are subject to deteriorating action by a number of factors, chiefly sunlight, fungi, and bacteria. These and others of lesser overall importance interact in a complicated pattern at a rate depending upon the nature of the fabric itself and upon the particular set of environmental circumstances attending the exposure. The nature of the past war was such that fabrics in immense quantity were exposed for long periods to the degrading influence of intense tropical sunlight, to high humidity and wetness which favored fungus action, or to actual drenching by rain and contact with soil thus introducing possible bacterial action into the picture; or in any given case a fabric may have been exposed alternately or concurrently to two or more such factors. The destructive action of fungi was especially marked in the Southwest Pacific and in the China-Burma-India region.

This soon led to an intense program of "tropic-proofing," first perhaps in Australia, to be followed closely by responsible organizations in the United States, Canada, England, India, and New Zealand. The work, at first largely empirical, became gradually more fundamental as the need for more knowledge concerning the basic nature and cause of the deterioration became increasingly apparent. Late in 1944, the Quartermaster Corps undertook a pro-

* Extract presented at the Meeting of the American Association for the Advancement of Science, Boston, Dec. 1946; abstract in Amer. Journ. Bot. 33: 832–833. 1946.

gram of basic research into the causes and processes involved in the deterioration of fabrics and like materials. The object of this program was to provide a basis for the production of field items better qualified to withstand such adverse conditions.

One of the first phases of the broad program was a study of the nature of the fungous flora of fabrics deteriorated in the tropics. Many hundreds of cultures were accumulated from a wide range of tropical regions. These came through a variety of channels, but the large majority of those included in the present paper were isolated at this laboratory during the winter of 1944–45 by Dr. Elwyn T. Reese. These are designated by "PQMD" preceding the number. Nearly all of them were made from samples from the Southwest Pacific or India. These had been sent from the field accompanied by certain essential data and wrapped to specification to preclude contamination in transit. Several isolation procedures were used in order to select not only those organisms capable of cellulolytic action but also a fair sampling of others present. In the aggregate the cultures that have been assembled have provided what may be considered a good background of knowledge of the mycobiota of tropical fabrics as a potentiality. An evaluation of the behavior and action and relative importance of each species in nature is another matter requiring laboratory work with complementing skillful field observation over a considerable period of time.

OBJECTIVES AND LIMITATIONS OF THE PRESENT PAPER

The purpose of this paper is to report the results of a series of routine assays for cellulolytic activity of some of the fungi isolated from military cotton fabrics and related materials which had undergone deterioration in the tropics. They were conducted for the purpose of separating the cellulolytic from the non-cellulolytic species and to provide a fair conception of the comparative rapidity of action. The results are to be considered preliminary and suggestive, an early step toward an understanding of the nature and activity of the mycobiota of fabrics and similar materials exposed under unfavorable conditions. Certain cultures have been selected for more intensive study and for use in the biochemical phases of the Quartermaster program.

A really meaningful comparison of the cellulolytic activity of strains or species or organisms can be made only after the particular set of cultural conditions optimal for each entity is known. A relatively full understanding of the requirements and behavior of an organism with regard to its cellulolytic activity in pure culture is essential if the organism is to be used in the experimental work of a program of fundamental research directed toward the development of biologically resistant materials. The establishment of a species as a "strong" or "weak" cellulose destroyer under laboratory conditions must not, however, be interpreted empirically as an index to the economic importance of the particular species in the deterioration of fabrics under conditions of field exposure. For example there is as yet no evidence that *Myrothecium verrucaria* (= *Metarrhizium glutinosum*), which perhaps is the most strongly cellulolytic species of fungus yet discovered, and therefore widely used in laboratory experimental work, is of any significance at all in the destruction of fabrics in the field.

Conversely the demonstration that the members of a species are incapable of direct enzymatic action against the basic cellulosic ingredients of a fabric does not mean that such a species may be dismissed as of no significance. Much depends upon the nature of the fabric and its intended use. In the destruction of waterproofing or various coating materials a fabric may be rendered unserviceable without direct cellulolytic action, or the way thus may be opened for the later cellulolytic attack. In this connection it may be noted that in the prewar years when the British Cotton Industry found itself faced with numerous claims for mildew damage to cotton goods in transit to Eastern markets it was found that the main problem was that of controlling fungi which grow in the starchy material used in sizing. Galloway (**2**), in a statement of the problems confronting the industry, listed the objectionable effects of fungi as (*1*) musty smell, (*2*) colored stains, (*3*) production of acids and alkali, and (*4*) tendering or actual enzymatic destruction of the fibers. Staining, which included both pigment production and the coloration of the fungus itself, was said to be by far the most important effect and the cause of the majority of damage claims.

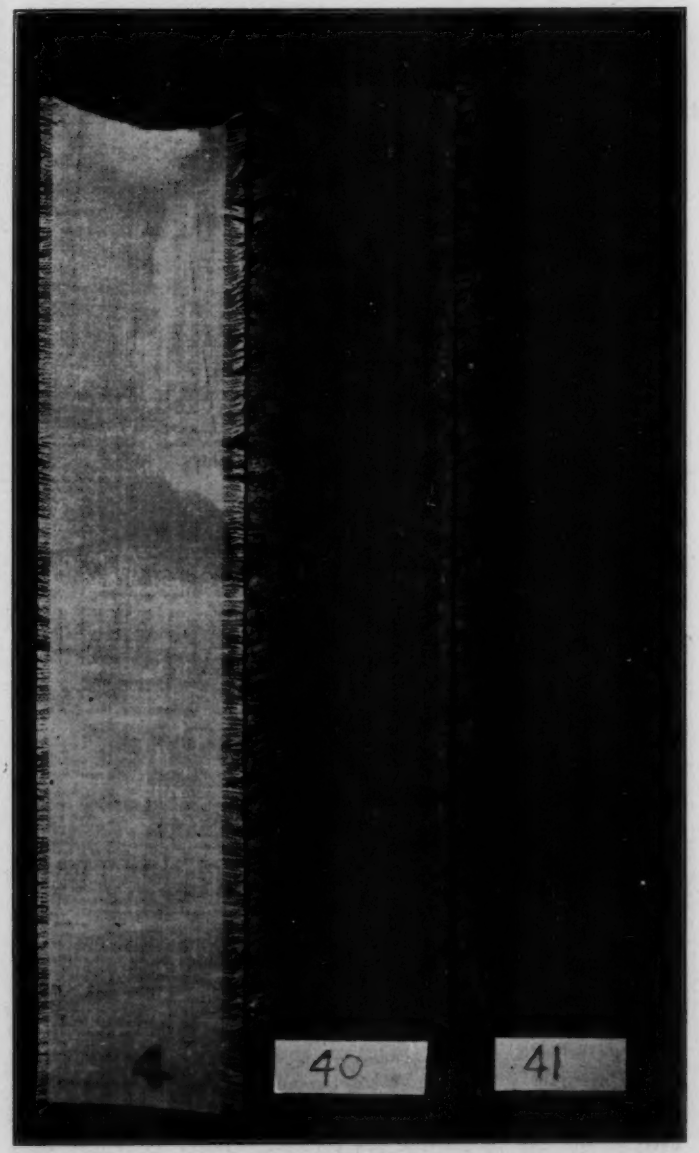

FIG. 1. Assay of cellulolytic activity.

Many organisms commonly isolated from fabrics are there only in the form of spores deposited with the dust of the atmosphere and are incapable of any direct action against the basic ingredients of the fabric, whereas others potentially destructive are probably seldom operative in nature. There is reason to believe that continued work will eventually demonstrate that a large portion of the actual decay of the fibers, especially under a defined range of environmental factors, is due to a relatively small number of species of fungi.

A review of pertinent literature will not be undertaken, for it would be of little value unless it could be more critical than space will permit in this paper. That appearing prior to 1927 was summarized by Thaysen and Bunker (13), and enough of the publications appearing since that time to provide an entrance to the literature are listed at the end of the present paper.

A more detailed analysis of the identity, distribution, specificity for materials, and economic significance of some of the species and groups of species should be forthcoming in future articles.

MATERIALS AND METHODS

The assays were conducted concurrently with other work extending over a period from October 1944 to December 1945. The basic method was that described recently by Greathouse, Klemme, and Barker (4) in which a spore suspension is placed on a strip of cotton fabric in the presence of a solution of mineral salts and incubated. The decline in tensile strength of the cloth was taken as a measure of the cellulolytic activity of the organism. The nutrient solution used was that designated as "Formula A" and had the following composition in grams per liter: K_2HPO_4, 1.3940; $MgSO_4 \cdot 7H_2O$, 0.7395; NH_4NO_3, 1.0006; $CaCO_3$, 0.005; NaCl, 0.005; $Fe_2(SO_4)_3 \cdot 7H_2O$, 0.001; $MnSO_4$, 0.001; and $ZnSO_4 \cdot 7H_2O$, 0.001. The initial pH was approximately 6.8.

In the course of the tests, several successive slight variations were introduced into the basic method. These are summarized briefly in Table I. When 16-oz. French square bottles were used (method variants 1–3), a 1½ inch diameter glass fabric insert was used in the cap to provide for aeration. Forty milliliters of a

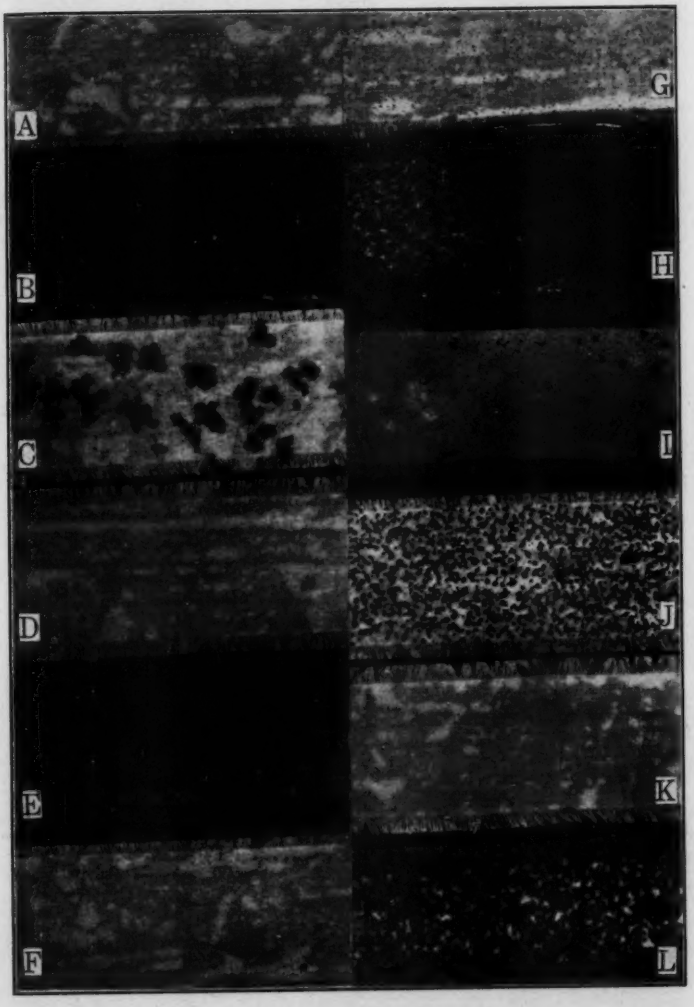

FIG. 2. Assay of cellulolytic activity.

medium consisting of the mineral salts plus 1.5 per cent agar was placed in each bottle, autoclaved for 20 minutes at 15 lbs. pressure, and the bottles laid on their sides to cool. The strips of fabric, cut to a length of approximately six inches and raveled to a width of exactly 1 inch, were subjected to similar autoclaving, dry, and

TABLE I

OUTLINE OF VARIATION IN BASIC METHOD USED FOR ASSAY FOR CELLULOLYTIC-ACTIVITY OF FUNGI

Variant	Cloth		Incubation	
	Type	Tensile Strength*	Vessel	Temp.-humidity
1	4 oz. sheeting	55.2 ± 0.5, $\sigma = 3.5$, $N = 87$*	Bottle	Room
2	3.3 oz. sheeting	38.2 ± 0.2, $\sigma = 1.6$, $N = 351$	Bottle	Room
3	3.3 oz. sheeting	38.2 ± 0.2, $\sigma = 1.6$, $N = 351$	Bottle	85° F., 80% r.h.
4	3.3 oz. sheeting	38.2 ± 0.2, $\sigma = 1.6$, $N = 351$	Test tube	85° F., 80% r.h.
5	Gray duck (a)	179.2 ± 2.1, $\sigma = 5.6$, $N = 9$	Bottle	85° F., 80% r.h.
6	Gray duck (a)	179.2 ± 2.1, $\sigma = 5.6$, $N = 9$	Test tube	85° F., 80% r.h.
7	Gray duck (b)	143.2 ± 1.5, $\sigma = 12.2$, $N = 65$	Test tube	85° F., 80% r.h.
8	12.29 oz. gray duck	169.3 ± 3.8, $\sigma = 15.0$, $N = 17$	Test tube	85° F., 80% r.h.

* Tensile strength is expressed as the arithmetic mean of a number of replicates (N) in pounds.

σ = Standard deviation $\frac{\sqrt{\Sigma X^2}}{N}$ where X is the deviation of the individual values from the mean.

in a separate container. They were then transferred under sterile conditions by means of a long pair of forceps one to each bottle and placed flat on the agar. Strips of 3.3 oz. bleached cotton sheeting incubated in accordance with this procedure are shown in figures 1 and 2. When test tubes were employed (method variants 4–8), the size used was 150 × 25 mm., of 75 ml. capacity. Twenty-five milliliters of the salts solution was placed in each and the cloth strip inserted. Slightly less than the lower half of the strip was submerged. The tubes were then plugged with cotton, sterilized as above, and allowed to cool. The method is illustrated in figure 3.

The variations in procedure outlined in Table I should be taken into consideration in interpreting the results. There is evidence that some organisms capable of attacking gray duck were, due

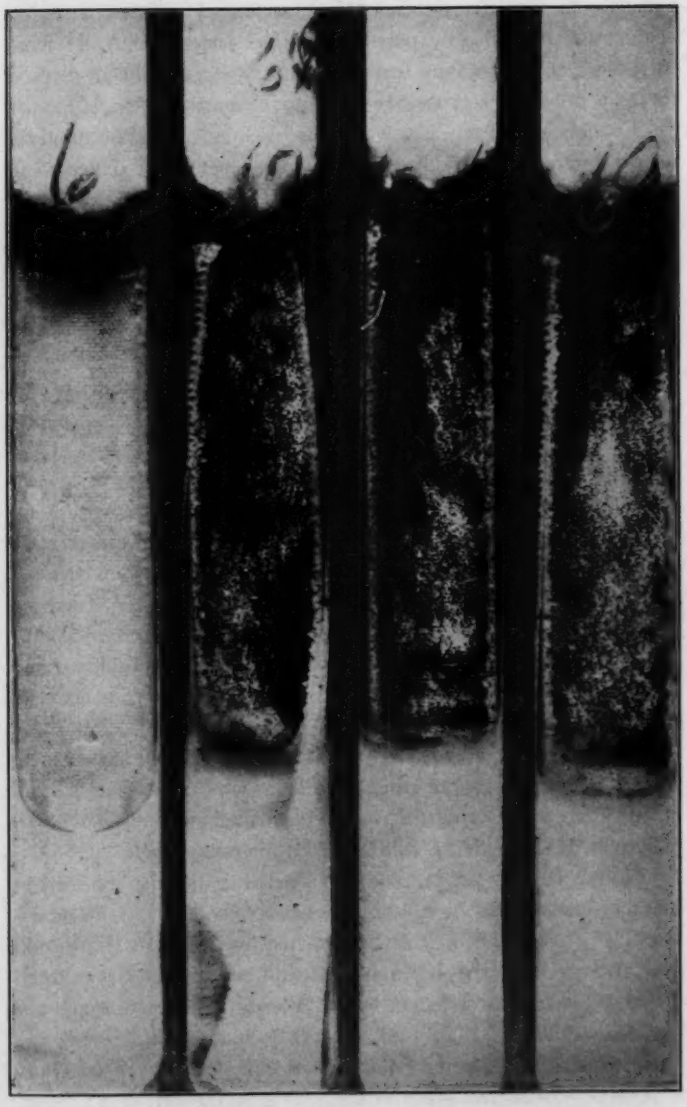

FIG. 3. *Aspergillus luchuensis* on gray cotton duck.

probably to nutritive deficiencies, incapable of cellulolytic action against the more highly purified bleached sheeting. Aside from this, there are few, if any instances where the variation in method meant a difference of positive or negative results.

The inoculum in all cases was prepared by growing the fungi on potato dextrose agar slants (20 g. dextrose per liter) under room conditions until good sporulation resulted. For most species a period of two to three weeks was found to be about right, although a few required a considerably longer period. The tube was then filled nearly to the top with sterile distilled water, the surface of the slant scraped with the tip of a 2 ml. pipette until a heavy spore suspension was obtained, and 2 ml. of the resulting suspension was distributed evenly over the surface of the cloth by means of the pipette.

When the bottle method was used the fungi were killed at the end of the incubation period by inverting the bottles, placing in each a few drops of formalin, replacing the ventilation caps with solid ones, and allowing to stand over night. When test tubes were used the strips were removed and washed in 70 per cent alcohol. In either case the strips were rinsed finally in tap water, dried for a few hours under room conditions, subjected to standard conditioning (24 hours at 70° F. and 65 per cent relative humidity), and broken on a motor driven Scott tester with a 3-inch space between the jaws.

About 20 cultures were assayed at a time. With each such lot were included two sets of controls, one on which was placed 2 ml. of distilled water, and the other inoculated with *Chaetomium globosum*, USDA *1042.4*, as a standard for comparison.

Culture numbers are preceded by a prefix indicating the institution at which the isolation was made, as follows:

PQMD. A series of isolations started by Prof. W. H. Weston at Harvard University in June 1944 and continued at the Philadelphia Quartermaster Depot by Dr. Elwyn T. Reese through the first half of 1945. Nearly all that are included in this paper are from items collected in the Southwest Pacific or India.

JQMD. Isolations made at the Jeffersonville (Indiana) Quartermaster Depot by, or under the direction of, Dr. G. W. Martin

or Dr. W. D. Gray from similar materials and over a similar period.

Fla. Isolations by Dr. G. F. Weber at University of Florida from Quartermaster items exposed experimentally at Camp Indian Bay, Florida.

Aust. Isolates made during the early period of the war in the Pacific by the Australian Mycological Panel, mostly from military items collected in New Guinea.

MIT. Isolates made during the early months of 1944 at the Chemical Warfare Service Development Laboratory, Massachusetts Institute of Technology, from items under test in local "tropical" chambers and from the tropics.

42nd Chem. Co. Isolates made in the Southwest Pacific by the 42nd Chemical Laboratory Co. of the Chemical Warfare Service in 1943 and sent to Chemical Warfare Service Development Laboratory, M.I.T.

USDA. Cultures obtained from the Division of Cotton and Other Fiber Crops and Diseases, Bureau of Plant Industry, U. S. Department of Agriculture—mostly of local origin.

RESULTS

The results are presented in Table II.

The figures cited for the inoculated strips are based on three to five replicates. The uninoculated controls from the several experiments were combined to arrive at the arithmetic means used as 100 per cent. Analysis of these figures indicates that the dispersion of the individual values around the arithmetic mean is quite large. Thus, for the 3.3 oz. cloth the 3σ limits are about 19 per cent and for the 4.0 oz. cloth about 12.5 per cent. In view of such wide dispersion and the fact that a maximum of five replicates were used for any given percentage figure, it is probably reasonable to assume that any decline in tensile strength less than 20 per cent is of doubtful significance.

TABLE II

ASSAY OF FUNGI FOR CELLULOLYTIC ACTIVITY

Species	Culture Number	Source[1]	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Absidia capillata van Tieg.	PQMD 8b	Undershirt	Bougainville Is.	3	99	102	105		0
[2] *Acladium* sp.?	PQMD 45f	Shoe	Ledo, India	2	42	31	18		4
[2] *Acladium* sp.	PQMD 45f	Shoe	Ledo, India	3	38	16	7		4
[2] *Acladium* sp.	PQMD 49c	Web belt	Ledo, India	2	52	39	31		4
Acremonium sp.	PQMD 1b	Tent	Bougainville Is.	1		37	24		3
Acremonium sp.	PQMD 1b	Tent	Bougainville Is.	7		41	25	17	4
Acremonium sp.	PQMD 121j	Tarpaulin	Panama	3	43	32	29		2
Alternaria tenuis auct.	PQMD 26a	Canvas	Russell Is.	2	94	79	65		3
Alternaria tenuis auct.	PQMD 73b	Plastic canteen	Finsch., N.G.	3	80	76	70		2
Alternaria tenuis auct.	Fla B-7	Tent	Florida	3	82	79	59		3
Alternaria tenuis auct.	Fla B-47	Canvas	Florida	3	76	66	59		2
Alternaria tenuis auct.	Fla B-48	Canvas	Florida	3	74	67	55		4
Arthrosporium sp.	PQMD 124f	Duck	Panama	3	108	106	101		0
Aspergillus Chevalieri (Mang.) Th. & Ch.	PQMD 52d	Wool-cotton shirt	Finsch., N.G.	2	104	106	113		0
Aspergillus Chevalieri (Mang.) Th. & Ch.	PQMD 53d	Leather strap	Finsch., N.G.	2	105	100	102		0
Aspergillus Chevalieri (Mang.) Th. & Ch.	PQMD 59c	Canvas kit	Finsch., N.G.	2	107	103	102		0

[1] Designation of item from which organism was isolated is shortened as much as possible; in all cases except where obvious or otherwise designated, the base material is cellulose.

[2] Brackets so used indicate that the numbers included represent but one species.

otherwise designated, the base material is cellulose.

[2] Brackets so used indicate that the numbers included represent but one species.

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Aspergillus Chevalieri (Mang.) Th. & Ch. (cont.)	PQMD 64c	Shirt	Finsch., N.G.	3	100	101	96		0
A. Chevalieri var. *intermedius* Th. & Raper	PQMD 58b	Leatherette	Finsch., N.G.	2	104	101	101		0
A. Fischeri Wehm.	Fla A-11		Florida	1		87	62	40	3
A. Fischeri Wehm.	Fla A-11		Florida	4	68			0	2
A. Fischeri Wehm.	Fla A-11		Florida	7	91			36	4
A. flavipes (Bain & Sart.) Th. & Ch.	PQMD 24a	Netting	Russell Is.	1	63	56	47		4
A. flavipes (Bain & Sart.) Th. & Ch.	Fla A-14		Florida	2	86	68	55		4
A. flavipes (Bain & Sart.) Th. & Ch.	Fla B-67	Leggings	Florida	3	91	75	69		4
A. flavus Link	PQMD 4m	Shoe	Bougainville Is.	1	103	95	93		1
A. flavus Link	PQMD 10e	Canvas	Oro Bay, N.G.	1		91	98	98	0
A. flavus Link	PQMD 13a	Canvas	Espiritu Santo Is.	1		92	101	91	1
A. flavus Link	PQMD 13c	Canvas	Espiritu Santo Is.	8		101	98	95	3
A. flavus Link	PQMD 45c	Shoe	Ledo, India	2	102	97	107		0
A. flavus Link	PQMD 48d	Tent	Ledo, India	2	102	105	99		1
A. flavus Link	PQMD 63c	Rope	Finsch., N.G.	2	110	105	97		0
A. flavus Link	PQMD 70a	Shoe	Finsch., N.G.	3	94	101	91		0
A. flavus Link	PQMD 73a	Plastic canteen	New Guinea	3	99	95	99		0
A. flavus Link	PQMD 91a	Canvas	Finsch., N.G.	6	113			112	1
A. flavus Link	PQMD 91a	Canvas	Finsch., N.G.	7	102			107	0

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
A. flavus Link (cont.)	JQMD 189	Tent	Hollandia, N.G.	3	105	97	95		1
A. flavus Link	Fla A-5		Florida	1		109	103	101	0
A. flavus Link	Fla A-6		Florida	1		105	98	111	1
A. flavus Link	Fla A-7			1		109	101	111	1
A. flavus Link	Fla B-20	Canteen cover	Florida	3	93	99	93		0
A. flavus Link	Aust 3	Shoe	New Guinea	3	100 +	96	91		0
A. fumigatus Fres.	PQMD 6b	Tent	Bougainville Is.	1		49	27	19	4
A. fumigatus Fres.	PQMD 6b	Tent	Bougainville Is.	4	81			16	3
A. fumigatus Fres.	PQMD 6b	Tent	Bougainville Is.	7	110			36	4
A. fumigatus Fres.	PQMD 45h	Shoe	Ledo, India	2	92	92	76		3
A. fumigatus Fres.	PQMD 45h	Shoe	Ledo, India	7		93		41	4
A. fumigatus Fres.	PQMD 45h	Shoe	Ledo, India	4		87		72	2
A. fumigatus Fres.	PQMD 91d	Canvas	Finsch., N.G.	3	84	63	44		4
A. fumigatus Fres.	JQMD 206	Tarpaulin	Cairo, Egypt	3	99	92	99		2
A. fumigatus Fres.	MIT 3c	Gas mask cover	India	3	57	26	16		4
A. fumigatus series	Fla B-21	Canteen cover	Florida	3	93	96	97		1
A. fumigatus series	Fla B-26	Cloth	Florida	3	96	97	89		1
A. japonicus Saito	PQMD 124g	Duck	Panama	3	100	95	96		0
A. luchuensis Inui	PQMD 23b	Rope	Port Moresby, N.G.	4		106		98	0
A. luchuensis Inui	PQMD 23b	Rope	Port Moresby, N.G.	7		108		89	3
A. luchuensis Inui	JQMD 745	Tent	Port Moresby, N.G.	4	92			97	0

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
A. luchuensis Inui (cont.)	JQMD 745	Tent	Port Moresby, N.G.	4				98	0
A. luchuensis Inui	JQMD 745	Tent	Port Moresby, N.G.	7				78	4
A. luchuensis Inui	JQMD 745	Tent	Port Moresby, N.G.	7			73		4
A. luchuensis Inui	JQMD 745	Tent	Port Moresby, N.G.	6			68		4
A. luchuensis Inui	JQMD 745	Tent	Port Moresby, N.G.	7	103			92	3
A. luchuensis Inui	JQMD 745	Tent	Port Moresby, N.G.	6	75			49[1]	4
A. luchuensis Inui	JQMD 745	Tent	Port Moresby, N.G.	5	94	82	77		3
A. nidulans (Eid.) Wint.	Fla A-13		Florida	4		93		72	2
A. nidulans (Eid.) Wint.	Fla A-13		Florida	7		105		106	2
A. niger group	PQMD 4j	Shoe	Bougainville Is.	1	106	93	90		1
A. niger group	PQMD 4j	Shoe	Bougainville Is.	4	92			102	1
A. niger group	PQMD 4j	Shoe	Bougainville Is.	5	107	98	102		2
A. niger group	PQMD 4j	Shoe	Bougainville Is.	7	107			115	1
A. niger group	PQMD 10c	Canvas	Oro Bay, N.G.	1	94	103	94		1
A. niger group	PQMD 10c	Canvas	Oro Bay, N.G.	4	94			105	0
A. niger group	PQMD 10c	Canvas	Oro Bay, N.G.	7	114			110	1
A. niger group	PQMD 10c	Canvas	Oro Bay, N.G.	5	103	102	105		2
A. niger group	PQMD 17g	Tent	Espiritu Santo Is.	1			94	100	1

[1] Figure 3.

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
A. niger group (cont.)	PQMD 17g	Tent	Espiritu Santo Is.	4	102			102	0
A. niger group	PQMD 17g	Tent	Espiritu Santo Is.	5	103	100	95		2
A. niger group	PQMD 17g	Tent	Espiritu Santo Is.	7	111			112	0
A. niger group	PQMD 22c	Tarpaulin	Russell Is.	4	86			102	0
A. niger group	PQMD 22c	Tarpaulin	Russell Is.	7	112			108	0
A. niger group	PQMD 23f	Tarpaulin	Espiritu Santo Is.	1	92	98	105		2
A. niger group	PQMD 23f	Tarpaulin	Espiritu Santo Is.	4		106		101	0
A. niger group	PQMD 23f	Tarpaulin	Espiritu Santo Is.	7		110		110	1
A. niger group	PQMD 24d	Netting	Russell Is.	4		103		111	0
A. niger group	PQMD 24d	Netting	Russell Is.	7		109		111	1
A. niger group	PQMD 25a	Shoe	Russell Is.	1	100	103	105		2
A. niger group	PQMD 25a	Shoe	Russell Is.	4		108		103	0
A. niger group	PQMD 25a	Shoe	Russell Is.	7		106		112	1
A. niger group	PQMD 26f	Canvas	Russell Is.	4		103		103	0
A. niger group	PQMD 26f	Canvas	Russell Is.	7		110		109	1
A. niger group	PQMD 30a	Shoe	Kauai, T.H.	2	99	99	99		1
A. niger group	PQMD 30a	Shoe	Kauai, T.H.	4		108		108	1
A. niger group	PQMD 30a	Shoe	Kauai, T.H.	7		106		108	1
A. niger group	PQMD 34c	Canteen cover	New Guinea	1	105	101[1]	105		2
A. niger group	PQMD 34c	Canteen cover	New Guinea	4		103		101	1
A. niger group	PQMD 34c	Canteen cover	New Guinea	7		108		112	1
A. niger group	PQMD 36b	Tarpaulin	New Guinea	2	107	102	105		1
A. niger group	PQMD 36b	Tarpaulin	New Guinea	4		101		108	0
A. niger group	PQMD 36b	Tarpaulin	New Guinea	7		103		103	1
A. niger group	PQMD 38b	Tentage	Karachi, India	2	102	105	102		0
A. niger group	PQMD 45d	Shoe	Ledo, India	2	94	99	102		1
A. niger group	PQMD 45d	Shoe	Ledo, India	4		103		101	0
A. niger group	PQMD 45d	Shoe	Ledo, India	7		104		99	2

[1] Figure 2*G*.

[1] Figure 2G.

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
A. niger group (cont.)	PQMD 50c	Rope	Hawaii	2	99	105	97		1
A. niger group	PQMD 50c	Rope	Hawaii	4		106		98	0
A. niger group	PQMD 50c	Rope	Hawaii	7		105		106	1
A. niger group	PQMD 51c	Tent	Hawaii	2	107	105	102		1
A. niger group	PQMD 64b	Shirt	Finsch., N.G.	2	101	101	97		0
A. niger group	PQMD 95b	Shoe	Florida	4	89			102	0
A. niger group	PQMD 95b	Shoe	Florida	7	111			114	1
A. niger group	PQMD 104b	Rubber boot	Finsch., N.G.	4	84			105	0
A. niger group	PQMD 104b	Rubber boot	Finsch., N.G.	7	111			108	0
A. niger group	PQMD 106f	Legging	Finsch., N.G.	4	97			102	0
A. niger group	PQMD 106f	Legging	Finsch., N.G.	7	112			109	1
A. niger group	PQMD 108d	Shoe	Florida	4	86			102	0
A. niger group	PQMD 108d	Shoe	Florida	7	107			106	1
A. niger group	Fla A-1a	Tent	Florida	1		103	94	103	1
A. niger group	Fla A-2		Florida	1		103	100	105	1
A. niger group	Fla B-27	Cloth	Florida	3	97	99	104		1
A. niger group	Fla B-55	Tent	Florida	3	96	101	99		1
A. niger group	Fla C-98	Typewriter ribbon	Florida	3	102	104	99		1
A. niger group	Aust 26	Electrical	Australia	7		98		101	1
A. niger group	Aust 26	Electrical	Australia	4		108		106	1
A. niger group	42nd Chem. 19b			4		101		108	0
A. niger group	42nd Chem. 19b			7		101		99	1
A. ochraceus Wilh.	PQMD 26b	Canvas	Russell Is.	2	99	105	97		1
A. ochraceus Wilh.	PQMD 26b	Canvas	Russell Is.	4		103		103	0
A. ochraceus Wilh.	PQMD 26b	Canvas	Russell Is.	7		105		98	1
A. ochraceus Wilh.	PQMD 58c	Leatherette	Finsch., N.G.	2	100	92	92		2

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
A. Oryzae (Ahl.) Cohn	PQMD 22b	Canvas	Russell Is.	4	86		102		0
A. Oryzae (Ahl.) Cohn	PQMD 22b	Canvas	Russell Is.	7	117		110		1
A. Oryzae (Ahl.) Cohn	42nd Chem. 18a	Gas mask cover	Milne Bay, N.G.	3	106	93	94		0
A. repens (Corda) DeBary	PQMD 44c	Tobacco	S.W. Pacific	2	105	99	102		0
A. repens (Corda) DeBary	PQMD 56f	Rope	Finsch., N.G.	2	102	100	104		1
A. repens (Corda) DeBary	PQMD 56f	Rope	Finsch., N.G.	3	101	98	94		0
A. repens (Corda) DeBary	PQMD 56f	Rope	Finsch., N.G.	7		99	103	97	0
A. repens (Corda) DeBary	PQMD 58h	Leatherette	Finsch., N.G.	2	102	102	99		0
A. repens (Corda) DeBary	PQMD 59g	Canvas	Finsch., N.G.	2	84	98	91		0
A. repens (Corda) DeBary	PQMD 59g	Canvas	Finsch., N.G.	3	84	97	91		0
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 4d	Shoe	Bougainville Is.	1		100	103		1
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 4d	Shoe	Bougainville Is.	1	110	102	103		1
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 4d	Shoe	Bougainville Is.	4		106		95	0
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 4d	Shoe	Bougainville Is.	7		101		109	1
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 26h			7		100	102	95	1
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 31c	Canvas	New Guinea	1	100	105	103		2
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 39a	Canvas	Karachi, India	2	99	102	102		1
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 41a	Rope	Karachi, India	2	107	107	105		0

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
A. Sydowi (Bain. & Sart.) Th. & Ch. (cont.)	PQMD 48e	Tent	Ledo, India	2	97	86	84		3
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 54a	Tent	Finsch., N.G.	2	104	104	102		0
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 55e	Leather	Finsch., N.G.	2	103		104		0
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 58d	Leatherette	Finsch., N.G.	2	106	101	106		2
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 58e	Leatherette	Finsch., N.G.	2	99	107	102		0
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 58g	Leatherette	Finsch., N.G.	2	109	99	99		0
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 58g	Leatherette	Finsch., N.G.	2	97	103	104		0
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 60a	Leatherette	Finsch., N.G.	2	109	100	105		1
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 61a		Finsch., N.G.	2	103	103	98		0
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 62d	Corncob pipe	Finsch., N.G.	2	100	103	99		0
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 62f	Corncob pipe	Finsch., N.G.	2	107	104	93		0
A. Sydowi (Bain. & Sart.) Th. & Ch.	PQMD 73c	Plastic canteen	Finsch., N.G.	3	96	93	94		0
A. Sydowi (Bain. & Sart.) Th. & Ch.	Fla B-18	Canteen cover	Florida	3	98	99	89		0
A. Sydowi (Bain. & Sart.) Th. & Ch.	Fla B-42	Canvas	Florida	3	88	103	100		0

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
A. Tamarii Kita	PQMD 9a	Cellophane	Oro Bay, N.G.	1		89	101	87	2
A. Tamarii Kita	PQMD 9a	Cellophane	Oro Bay, N.G.	4	105			102	0
A. Tamarii Kita	PQMD 9a	Cellophane	Oro Bay, N.G.	7	108			110	2
A. Tamarii Kita	PQMD 50b	Rope	Hawaii	2	99	105	105		1
A. Tamarii Kita	PQMD 51h	Tent	Hawaii	2	107	105	94		1
A. terreus Thom	PQMD 72f	Leather	Finsch., N.G.	3	67	42	32		4
A. terreus Thom	PQMD 91c	Canvas	Finsch., N.G.	4	72			19	4
A. terreus Thom	PQMD 91c	Canvas	Finsch., N.G.	7	99			42	4
A. terreus Thom	PQMD 94b	Trousers	Finsch., N.G.	7			23		4
A. terreus Thom	PQMD 94b	Trousers	Finsch., N.G.	7			22		4
A. terreus Thom	PQMD 106g	Legging	Finsch., N.G.	4	47			16	3
A. terreus Thom	PQMD 106g	Legging	Finsch., N.G.	7	94			43	3
A. terreus Thom	JQMD 481	Cord	Jeffersonville, Ind.	3	63	41	26		4
A. terreus Thom	Fla B-19	Canteen cover	Florida	3	70	51	34		4
A. terreus Thom	MIT 7	Wood	Cambridge, Mass.	2	75	58	41		3
A. unguis (Em.-Weil & Gaud.) Th. & Rap.	PQMD 8f	Undershirt	Oro Bay, N.G.	1		103	96	96	1
A. unguis (Em.-Weil & Gaud.) Th. & Rap.	PQMD 8f	Undershirt	Oro Bay, N.G.	1		87	103	92	1
A. unguis (Em.-Weil & Gaud.) Th. & Rap.	PQMD 25b	Shoe	Russell Is.	1		103	96	100	1
A. unguis (Em.-Weil & Gaud.) Th. & Rap.	PQMD 30b	Shoe	Kauai, T.H.	2	97	105	107		0
A. unguis (Em.-Weil & Gaud.) Th. & Rap.	PQMD 45e	Shoe	Ledo, India	2	97	105	105		0

TABLE II—*continued*

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
A. unguis (Em.-Weil & Gaud.) Th. & Rap. (cont.)	PQMD 53c	Leather	Finsch., N.G.	2	110	100	103		0
A. unguis (Em.-Weil & Gaud.) Th. & Rap.	PQMD 64a	Shirt	Finsch., N.G.	2	102	98	100		0
A. unguis (Em.-Weil & Gaud.) Th. & Rap.	PQMD 65b	Canvas	Finsch., N.G.	2	99	102	94		0
A. unguis (Em.-Weil & Gaud.) Th. & Rap.	PQMD 102g	Canvas	Finsch., N.G.	4	90			98	0
A. unguis (Em.-Weil & Gaud.) Th. & Rap.	PQMD 102g	Canvas	Finsch., N.G.	7	110			109	0
A. ustus (Bain) Th. & Ch.	PQMD 4h	Shoe	Bougainville Is.	1	96	102	93		2
A. ustus (Bain) Th. & Ch.	PQMD 4h	Shoe	Bougainville Is.	4		72		41	1
A. ustus (Bain) Th. & Ch.	PQMD 4h	Shoe	Bougainville Is.	7		103		84	3
A. ustus (Bain) Th. & Ch.	PQMD 51g	Tent	Hawaii	2	107	105	107		1
A. ustus (Bain) Th. & Ch.	PQMD 57c	Imitation leather	Finsch., N.G.	2	104	93	89		3
A. ustus (Bain) Th. & Ch.	JQMD 272			3	93	93	93		2
A. ustus (Bain) Th. & Ch.	Fla A-12		Florida	1		100	83	63	4
A. ustus (Bain) Th. & Ch.	Aust 22	Silk	New Guinea	3	90	70	69		3
A. ustus var. *laevis* (Block.) Th. & Rap.	PQMD 24a-2	Netting	Russell Is.	1	100	96	98		3
A. ustus var. *laevis* (Block.) Th. & Rap.	PQMD 24a-2	Netting	Russell Is.	3	97	96	96		1
A. ustus var. *laevis* (Block.) Th. & Rap.	PQMD 24a-2	Netting	Russell Is.	4		82		53	2
A. ustus var. *laevis* (Block.) Th. & Rap.	PQMD 24a-2	Netting	Russell Is.	7		102		101	2
A. ustus var. *laevis* (Block.) Th. & Rap.	PQMD 24a-2	Netting	Russell Is.	7			96	101	2

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
A. versicolor (Vuill.) Tira.	PQMD 4i	Shoe	Bougainville Is.	1	106	86	91		1
A. versicolor (Vuill.) Tita.	PQMD 17d	Tent	Espiritu Santo Is.	1			94	100	1
A. versicolor (Vuill.) Tira.	PQMD 26d	Canvas	Russell Is.	2	103		101	98	2
A. versicolor (Vuill.) Tira.	PQMD 26e	Canvas	Russell Is.	1	101		101	101	3
A. versicolor (Vuill.) Tira.	Fla C-18	Canteen bag	Florida	3		103	105		1
A. versicolor (Vuill.) Tira.	Fla C-18	Canteen bag	Florida	3			97		2
A. versicolor (Vuill.) Tira.	MIT 1c	Cellophane	India	4		108		106	1
A. versicolor (Vuill.) Tira.	MIT 1c	Cellophane	India	7		101		107	1
A. Wentii Weh.	PQMD 44a	Tobacco	S.W. Pacific	1	107	99	105		1
A. Wentii Weh.	PQMD 44a	Tobacco	S.W. Pacific	7		104	99	95	1
Aspergillus sp.	PQMD 62c	Corncob pipe	Finsch., N.G.	2	102	104	100		0
Aspergillus sp.	PQMD 62e	Corncob pipe	Finsch., N.G.	2	97	103	94		0
Botryodiplodia Theobromae Pat.	PQMD 2b	Duck	Bougainville Is.	1	102	97	97		1
Botryodiplodia Theobromae Pat.	PQMD 59d	Canvas	Finsch., N.G.	3	91	88	93		1
Botryodiplodia Theobromae Pat.	PQMD 67a	Trousers	Finsch., N.G.	7		92	73	83	3
Brachysporium sp.	PQMD 18c	Tent	Espiritu Santo Is.	1	54	38	42		1
Brachysporium sp.	PQMD 18c	Tent	Karachi, India	2	92	47	24		2
Brachysporium sp.	PQMD 38d	Tent	Karachi, India	2	79	73	55		3
Brachysporium sp.	PQMD 63b	Rope	Finsch., N.G.	2	84	76	55		0
Brachysporium sp.	PQMD 70g	Shoe	Finsch., N.G.	3	45	24	23		3
Brachysporium sp.	Fla B-57	Tent	Finsch., N.G.	3	72	59	44		4
Brachysporium sp.	Fla B-57	Tent	Finsch., N.G.	3	70	47	28		4

TABLE II—*continued*

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Cephalosporium sp.	Fla B-5	Tent	Florida	3	50	25	14		1
Cephalosporium sp.	Fla B-5	Tent	Florida	3	47	27	15		2
Cephalothecium roseum Corda	Fla B-50	Canvas	Florida	3	103	95	102		1
Cephalothecium roseum Corda	Fla B-50	Canvas	Florida	3	89	97	93		1
Cephalothecium sp.?	PQMD 124d	Duck	Panama	3	32	23	16		3
Chaetomella sp.	PQMD 40c	Canvas	Karachi, India	2	89	68	52		3
Chaetomium funicolum Cooke	PQMD 33c	Belt	New Guinea	1	13	0[1]			4
Chaetomium funicolum Cooke	PQMD 34d	Canteen cover	New Guinea	1	11	0[2]			4
Chaetomium funicolum Cooke	PQMD 35e	Tent	New Guinea	2	0				4
Chaetomium funicolum Cooke	PQMD 36d	Tarpaulin	New Guinea	2	18	0			4
Chaetomium funicolum Cooke	PQMD 42a	Cap	Chabua, India	2	13	0			4
Chaetomium funicolum Cooke	Fla B-12	Tent	Florida	3	16	0			4
Chaetomium funicolum Cooke	Aust 12	Seed	Australia	3	25	13	7		3
C. globosum Kunze	PQMD 32b	Tent	New Guinea	1	18	0[3]			4
C. globosum Kunze	PQMD 38f	Tent	Karachi, India	2	16	0			4
C. globosum Kunze	PQMD 59h	Canvas	Finsch., N.G.	2	18	0			4
C. globosum Kunze	PQMD 71g	Glove	Finsch., N.G.	3	8	0			4
C. globosum Kunze	Fla C-12	Shower curtain	Florida	3	12	0			4
C. globosum Kunze	USDA 1042.4			1	13	0[4]			4
C. globosum Kunze	USDA 1042.4			3	5	0			4
C. globosum Kunze	USDA 1042.4			4	46			0	4

[1] Figure 2*E*. [2] Figure 2*H*. [3] Figure 2*B*. [4] Figure 2*L*.

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
C. globosum Kunze (cont.)	USDA 1042.4			6	21	0			4
C. globosum Kunze	USDA 1042.4			7	42			0	4
C. globosum Kunze	USDA 1042.4			7		7	0		4
C. globosum Kunze	USDA 1042.4			5	42	27	10		4
C. indicum Corda	PQMD 46b	Rope	Ledo, India	2	26	0			4
C. indicum Corda	PQMD 47c	Wax paper	Ledo, India	2	26	0			4
Cladosporium herbarum (Pers.) Link	PQMD 17b	Tent	Espiritu Santo Is.	1		101	100		0
Cladosporium herbarum (Pers.) Link	PQMD 17e	Tent	Espiritu Santo Is.	1		80	67	49	3
Cladosporium herbarum (Pers.) Link	PQMD 17e	Tent	Espiritu Santo Is.	2	93	82	85		2
Cladosporium herbarum (Pers.) Link	PQMD 49a	Web belt	Ledo, India	2	78	69	52		4
Cladosporium herbarum (Pers.) Link	PQMD 52a	Shirt	Finsch., N.G.	2	112	103	106		1
Cladosporium herbarum (Pers.) Link	PQMD 55b	Leather	Finsch., N.G.	2	82	71	37		2
Cladosporium herbarum (Pers.) Link	PQMD 71b	Glove	Finsch., N.G.	3	87	86	74		1
Cladosporium herbarum (Pers.) Link	PQMD 119g	Canvas	Panama	3	94	91	95		1
Cladosporium herbarum (Pers.) Link	PQMD 120g	Tarpaulin	Panama	3	101	87	87		1
Cladosporium herbarum (Pers.) Link	PQMD 121e	Tarpaulin	Panama	3	103	102	101		1
Cladosporium herbarum (Pers.) Link	PQMD 121k	Tarpaulin	Panama	3	94	78	72		2

TABLE II—*continued*

Cladosporium herbarum (Pers.) Link	PQMD 121k	Tarpaulin	Panama	3	94	78	72		2

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Cladosporium herbarum (Pers.) Link (cont.)	PQMD 122c	Duck	Panama	3	87	85	76		2
Cladosporium herbarum (Pers.) Link	PQMD 122e	Duck	Panama	3	79	71			3
Cladosporium herbarum (Pers.) Link	Fla B-2	Tent	Florida	3	100	94	88		1
Cladosporium herbarum (Pers.) Link	Fla B-22	Canteen cover	Florida	3	96	90	93		0
Cladosporium herbarum (Pers.) Link	Fla B-28	Cloth	Florida	3	81	56	36		4
Cladosporium herbarum (Pers.) Link	Fla B-28	Cloth	Florida	3	62	38	26		4
Cladosporium herbarum (Pers.) Link	Fla B-29	Cloth	Florida	3	80	69	55		4
Cladosporium herbarum (Pers.) Link	Fla B-29	Cloth	Florida	3	75	53	35		4
Cladosporium herbarum (Pers.) Link	Fla B-43	Canvas	Florida	3	101	96	92		1
Cladosporium herbarum (Pers.) Link	Fla B-68	Leggings	Florida	3	92	71	68		2
Cladosporium herbarum (Pers.) Link	Fla C-42	Pistol cover	Florida	3	87	82	67		3
Cladosporium herbarum (Pers.) Link	Fla C-44	Pistol cover	Florida	3	92	72	63		3
Cladosporium herbarum (Pers.) Link	Fla C-4	Web belt	Florida	3	103	102	102		1
Cladosporium herbarum (Pers.) Link	Fla C-21	Canteen bag	Florida	3	106	108	105		0
Coniothyrium sp.	PQMD 121f	Tarpaulin	Panama	3	46	34	25		4

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Cunninghamella echinulata Thaxt.	PQMD 35c	Tent	New Guinea	1	107	111[1]	107		0
Curvularia brachyspora Boed.	Fla A-19		Florida	1		56	58	45	3
Curvularia brachyspora Boed.	Fla B-8	Tent	Florida	3	51	20[2]	11		4
Curvularia brachyspora Boed.	Fla B-9	Tent	Florida	3	57	30	13		4
Curvularia brachyspora Boed.	Fla B-46	Canvas	Florida	3	38	13	5		4
C. falcata (Teh.) Boed.	PQMD 120h	Tarpaulin	Panama	3	88	81	57		3
C. lunata (Wakk.) Boed.	PQMD 10d	Canvas	Oro Bay, N.G.	2	56	27	22		4
C. lunata (Wakk.) Boed.	PQMD 34b	Canteen cover	New Guinea	2	52	18	0		4
C. lunata (Wakk.) Boed.	PQMD 35d	Tent	New Guinea	1	60	47	33		4
C. lunata (Wakk.) Boed.	PQMD 120L	Tarpaulin	Panama	3	47	21	15		4
C. lunata (Wakk.) Boed.	PQMD 120L	Tarpaulin	Panama	3	65	21	8		4
C. lunata (Wakk.) Boed.	PQMD 121d	Tarpaulin	Panama	3	62	37	23		4
C. lunata (Wakk.) Boed.	JQMD 492	Socks	Hollandia, N.G.	3	34	19	13		4
Fusarium javanicum Koord.	PQMD 23d	Rope	Russell Is.	1	47	40	33		3
F. lateritium Nees	PQMD 120d	Tarpaulin	Panama	3	51	34	31		4
F. lateritium Nees	PQMD 120d	Tarpaulin	Panama	7		27	15	10	1
F. moniliforme Sheld.	Fla B-13	Tent	Florida	3	57	44	36		4
F. oxysporum Schl.	PQMD 23a	Rope	Russell Is.	1	94	92	98		0
F. oxysporum Schl.	PQMD 23c	Rope	Russell Is.	1	54	43	40		3

[1] Figure 2*K*. [2] Figure 1.

[1] Figure 2*K*. [2] Figure 1.

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
F. oxysporum Schl. (cont.)	PQMD 23e	Rope	Russell Is.	1	49	51	40		3
F. oxysporum Schl.	PQMD 23h	Rope	Russell Is.	1	54	49	38		1
F. oxysporum Schl.	PQMD 47e	Wax paper	Ledo, India	2	67	56	51		1
F. oxysporum Schl.	Fla B-61	Tent	Florida	3	39	27	19		4
F. oxysporum Schl.	Fla B-63	Tent	Florida	3	41	29	22		4
F. oxysporum Schl.	Fla C-8	Web belt	Florida	3	49	36	30		3
F. roseum Link	PQMD 38a	Tent	Karachi, India	2	93	77	66		2
F. roseum Link	PQMD 38e	Tent	Karachi, India	2	101	95	98		0
F. roseum Link	PQMD 38h	Tent	Karachi, India	2	103	95	90		0
F. roseum Link	PQMD 38g	Tent	Karachi, India	2	106	95	103		0
F. roseum Link	PQMD 40a	Canvas	Karachi, India	2	87	66	59		3
F. roseum Link	PQMD 125c	Canvas	Karachi, India	3	91	87	81		0
F. roseum Link	Fla A-17		Florida	1		76	53	49	4
F. roseum Link	Fla B-15	Tent	Florida	3	83	71	65		3
F. roseum Link	Fla B-24	Canteen cover	Florida	3	70	54	44		3
F. Scirpi var. *longipes* (W. & R.) W.	PQMD 23g	Rope	Russell Is.	1	58	53	38		2
F. Scirpi var. *longipes* (W. & R.) W.	PQMD 50f	Rope	Hawaii	2	101	101	102		0
F. semitectum Berk. & Rav.	PQMD 66b	Web strap	Finsch., N.G.	2	78	71	54		2
F. semitectum Berk. & Rav.	PQMD 122a	Duck	Panama	3	73	64	56		2
F. semitectum var. *majus* (Berk. & Lan.) Woll.	PQMD 121c	Tarpaulin	Panama	3		85	74		1
F. Solani (Mar.) Appel & Woll.	PQMD 21d	Tarpaulin	Espiritu Santo Is.	1	92	71	56		1

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
F. Solani (Mar.) Appel & Woll. (cont.)	PQMD 21f	Tarpaulin	Espiritu Santo Is.	1	49	33	38		3
F. Solani (Mar.) Appel & Woll.	PQMD 119d	Canvas	Panama	3	85	71	57		1
F. Solani (Mar.) Appel & Woll.	PQMD 123h	Duck	Panama	3	53	35	27		1
F. Solani (Mar.) Appel & Woll.	PQMD 125b	Duck	Panama	3	42	30	25		1
Fusarium sp. (sect. elegans)	PQMD 125d	Duck	Panama	3	97	93	79		2
Fusarium sp.	PQMD 22a	Canvas	Russell Is.	1	58	43	33		4
Fusarium sp.	PQMD 51d	Tent	Hawaii	2	98	100	99		0
Fusarium sp.	Fla B-14	Tent	Florida	3	87	77	75		0
Gliocladium roseum Bain.	Fla B-34	Cloth	Florida	3	70	56	47		1
Gliocladium sp.	PQMD 3a	Rope	Bougainville Is.	1		25	17		4
Gliocladium sp.	PQMD 3a	Rope	Bougainville Is.	7			90		1
Gliocladium sp.	PQMD 116b	Raincoat	Florida	3	45	17	6		4
Gliocladium sp.	PQMD 170		India	8	93		81	60	2
Gliocladium sp.	JQMD 316	Belt	Hollandia, N.G.	3	99	98	89		1
Gliocladium sp.	Fla A-3		Florida	1		94	91	85	3
Gliocladium sp.	Fla A-3		Florida	8		102	104	103	1
Gliocladium sp.	USDA T-1			3	98	95	91		0
Gliocladium sp.	USDA T-1			3	101	105	97		0
Gliocladium sp.	42nd Chem. Co. No. 23a	String	Milne Bay, N.G.	2	103	98	90		1

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Gliomastix convoluta (Harz) Mason	PQMD 2c	Duck	Bougainville Is.	1		14	0		4
Gliomastix convoluta (Harz) Mason	PQMD 2c	Duck	Bougainville Is.	1			0		4
Gliomastix convoluta (Harz) Mason	PQMD 2c	Duck	Bougainville Is.	1			0		4
Gliomastix convoluta (Harz) Mason	PQMD 4c	Shoe thread	Bougainville Is.	1		15	0		4
Gliomastix convoluta (Harz) Mason	PQMD 4c	Shoe thread	Bougainville Is.	8		51	22	13	4
Gliomastix convoluta (Harz) Mason	PQMD 4c	Shoe thread	Bougainville Is.	7	54	28	13		4
Gliomastix convoluta (Harz) Mason	PQMD 4c	Shoe thread	Bougainville Is.	5	29	0			4
Gliomastix convoluta (Harz) Mason	PQMD 124c	Duck	Panama	8		50	15	13	4
Gliomastix convoluta (Harz) Mason	Fla F-108		Florida	8		50	25	21	4
Gliomastix convoluta (Harz) Mason	Fla F-121		Florida	3	45	12	0		4
Gliomastix convoluta (Harz) Mason	Fla F-121		Florida	8		16	9	0	4
Gliomastix convoluta (Harz) Mason	Fla F-122		Florida	8		43	15	8	4
Gliomastix convoluta (Harz) Mason	Fla F-177		Florida	8		61	35	12	4
Gliomastix convoluta (Harz) Mason	MIT 16	Webbing	S.W. Pacific	3	27	9	8		4
Gliomastix convoluta (Harz) Mason	MIT 16	Webbing	S.W. Pacific	8		31	11	7	4

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Humicola sp.	PQMD 34e	Canteen cover	New Guinea	1	17	7[1]	0		4
Humicola sp.	PQMD 34e	Canteen cover	New Guinea	3	25	18	4		4
Humicola sp.	PQMD 73d	Plastic canteen	Finsch., N.G.	3	15	3	0		4
Humicola sp.	PQMD 175	Human ear	Texas	3	30	23	23		4
Humicola sp.	PQMD 68f	Rubber coat	Finsch., N.G.	3	77	63	52		3
Humicola sp.	PQMD 71c	Glove	Finsch., N.G.	3	88	73	62		4
Humicola sp.	PQMD 71e	Glove	Finsch., N.G.	3	74	64	56		1
Humicola sp.	PQMD 71f	Glove	Finsch., N.G.	3	73	52	55		3
Lichthemia Regnieri (Luc. & Cost.) Vuill.	PQMD 45b	Shoe	Ledo, India	2	103	98	101		0
Memnoniella echinata Riv. (Gallow)	PQMD 1c	Canvas	Bougainville Is.	1		26	13		3
Memnoniella echinata Riv. (Gallow)	PQMD 1c	Canvas	Bougainville Is.	7		17	3	2	4
Memnoniella echinata Riv. (Gallow)	PQMD 50d	Rope	Hawaii	2	87	73	47		0
Memnoniella echinata Riv. (Gallow)	PQMD 50g	Rope	Hawaii	2	29	24	20		3
Memnoniella echinata Riv. (Gallow)	PQMD 51a	Tent	Hawaii	2	41	26	16		4
Memnoniella echinata Riv. (Gallow)	PQMD 51e	Tent	Hawaii	2	33	26	16		4
Memnoniella echinata Riv. (Gallow)	PQMD 72i	Leather	Finsch., N.G.	3	64	50	28		0
Memnoniella echinata Riv. (Gallow)	PQMD 82d	Haversack	Finsch., N.G.	3	48	36	32		0

[1] Figure 2*I*.

[1] Figure 2*I*.

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Memnoniella echinata Riv. (Gallow) (cont.)	JQMD 177	Duck	Jeffersonville, Ind.	3	78	65	46		0
Memnoniella echinata Riv. (Gallow)	Fla B-32	Cloth	Florida	3	66	50	38		0
Memnoniella echinata Riv. (Gallow)	Fla B-66	Leggings	Florida	3	31	20	10		4
Memnoniella echinata Riv. (Gallow)	Aust 86		Florida	3	59	41	25		0
Metarrhizium brunneum Petch	PQMD 191[1]	Wireworms	Oregon	8	100		100	99	1
M. Anisopliae (Metsch.) Sorok.	PQMD 192[2]	Wireworms	Idaho	8	103		106	100	1
Microsporum gypseum (Bod.) Gui. & Grig.	PQMD 196	Wool fabric	Philadelphia	8	102		105	105	1
Mucor fumosus Naum.	MIT 3b	Gas mask cover	Mass.	3	104	99	96		0
Myrothecium roridum Tode ex Fr.	PQMD 201	*Aselepias* sp.	W. Africa	8	39	13	0		2
Myrothecium roridum Tode ex Fr.	PQMD 202	*Viola* sp.	England	8	35	8	0		2
Myrothecium roridum Tode ex Fr.	Fla B-11	Tent	Florida	3	0				4

[1] NRRL 1944. [2] NRRL 1945.

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
M. verrucaria (Alb. & Schw.) Ditm. ex Fr.	PQMD 34f	Canteen cover	New Guinea	1	17	11[1]	7		4
M. verrucaria (Alb. & Schw.) Ditm. ex Fr.	PQMD 70h	Shoe	Finsch., N.G.	3	0				4
M. verrucaria (Alb. & Schw.) Ditm. ex Fr.	PQMD 203	Potato	Cypress	8	10	0	0		3
M. verrucaria (Alb. & Schw.) Ditm. ex Fr.	PQMD 204	Citrus fruits	S. Rhodesia	8	40	27	15		3
M. inundatum Tode ex Fr.	PQMD 206	*Russula adusta*	England	8	106		96	92	2
Myxosporium sp.?	PQMD 120f	Tarpaulin	Panama	3	44	16	7		4
Oospora lactis (Fres.) Lindau	PQMD 59f	Canvas	Finsch., N.G.	3	52	38	24		0
Paecilomyces varioti Bain.	PQMD 10a	Canvas	Oro Bay, N.G.	1		92	94	96	0
Paecilomyces varioti Bain.	PQMD 21a	Tarpaulin	Espiritu Santo Is.	2	106	108	103		0
Paecilomyces varioti Bain.	PQMD 42c	Cap	Chabua, India	2	108	106	103		0
Paecilomyces varioti Bain.	PQMD 47d	Wax paper	Ledo, India	2	105	105	97		0
Paecilomyces varioti Bain.	PQMD 72e	Leather	Finsch., N.G.	3	101	98	98		0
Paecilomyces varioti Bain.	PQMD 82f	Haversack	Finsch., N.G.	3	103	104	109		0
Paecilomyces varioti Bain.	PQMD 108e	Shoe	Florida	7		98	99	97	1
Paecilomyces varioti Bain.	PQMD 50e	Rope	Hawaii	2	108	103	109		0
Paecilomyces varioti Bain.	Fla B-6	Tent	Florida	3	103	102	97		0
Paecilomyces varioti Bain.	Fla B-6	Tent	Florida	3	108	95	100		0
Penicillium Biourgeianum Zal.	PQMD 1a	Canvas	Bougainville Is.	1		103	104		1

[1] Figure 2*J*.

[1] Figure 2*J*.

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Penicillium Biourgeianum Zal. (cont.)	PQMD 1a	Canvas	Bougainville Is.	7		101	100	100	1
P. chrysogenum Thom	PQMD 51b	Tent	Hawaii	2	103	109	104		0
P. chrysogenum Thom	PQMD 60b	Rifle sling	Finsch., N.G.	2	106	107	105		0
P. citrinum series	PQMD 2a	Duck	Bougainville Is.	1		107	91		1
P. citrinum series	PQMD 4a	Shoe	Bougainville Is.	1		98	93		2
P. citrinum series	PQMD 4a	Shoe	Bougainville Is.	8		92	77	65	4
P. citrinum series	PQMD 4a	Shoe	Bougainville Is.	8		96	82	70	4
P. citrinum series	PQMD 4 *l*	Shoe lace	Bougainville Is.	1	98	96	91		2
P. citrinum series	PQMD 4 *l*	Shoe lace	Bougainville Is.	8		87	89	92	3
P. citrinum series	PQMD 4 *o*	Shoe	Bougainville Is.	1		94	92	87	2
P. citrinum series	PQMD 4 *o*	Shoe	Bougainville Is.	1		91	91	87	2
P. citrinum series	PQMD 7a	Raincoat	Bougainville Is.	1		100	96	91	2
P. citrinum series	PQMD 7a	Raincoat	Bougainville Is.	2		94	89	85	1
P. citrinum series	PQMD 7d	Raincoat	Bougainville Is.	1	98	100	98		2
P. citrinum series	PQMD 8a	Undershirt	Bougainville Is.	1		107	98	98	1
P. citrinum series	PQMD 8c	Undershirt	Bougainville Is.	1		83	89	78	2
P. citrinum series	PQMD 8g	Undershirt	Oro Bay, N.G.	1		83	89	77	2
P. citrinum series	PQMD 10b	Canvas	Oro Bay, N.G.	1		87	92	72	2
P. citrinum series	PQMD 18d	Canvas	Oro Bay, N.G.	1	103	101	101		1
P. citrinum series	PQMD 36c	Tarpaulin	New Guinea	2	103	98	101		0
P. citrinum series	PQMD 59b	Canvas	Finsch., N.G.	2	104	92	103		1
P. citrinum series	PQMD 62a	Corn cob pipe	Finsch., N.G.	2	102	98	97		0
P. citrinum series	PQMD 69a	Canvas	Finsch., N.G.	3	96	99	100		0
P. citrinum series	PQMD 69b	Canvas	Finsch., N.G.	3	93	95	89		1
P. citrinum series	JQMD 489	Shoe	Hollandia, N.G.	3	103	104	99		1
P. citrinum series	JQMD 489	Shoe	Hollandia, N.G.	5	99		100		2

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
P. corylophylum (near)	PQMD 42b	Cap	Chabua, India	2	108	101	106		0
P. cyaneum (Bain. & Sart.) Biourge	PQMD 4k	Shoe	Bougainville Is.	1	103	98	85		1
P. cyaneum (Bain. & Sart.) Biourge	PQMD 7b	Raincoat	Bougainville Is.	1		100	93	96	0
P. cyaneum (Bain. & Sart.) Biourge	PQMD 7c	Raincoat	Bougainville Is.	1		101	96	98	0
P. cyaneum (Bain. & Sart.) Biourge	PQMD 53b	Leather	Finsch., N.G.	2	107	110	104		0
P. Herquei Bain & Sart.	PQMD 124a-1	Duck	Panama	3	105	101	97		0
P. implicatum series	PQMD 45a	Shoe	Ledo, India	2	103	106	106		0
P. lilacinum Thom	PQMD 4e	Shoe	Bougainville Is.	1	102	100	96		2
P. luteum series (non-ascosporic)	PQMD 17c	Tent	Espiritu Santo Is.	8		102	104	102	1
P. luteum series (non-ascosporic)	PQMD 28b	Tent	Kauai, T.H.	2	101	103	98		0
P. luteum series (non-ascosporic)	PQMD 74d	Leather glove	Finsch., N.G.	3	92	90	76		3
P. luteum series (non-ascosporic)	PQMD 74d	Leather glove	Finsch., N.G.	3	86	87	79		2
P. luteum series (non-ascosporic)	JQMD 396	Blanket	Hollandia, N.G.	3	65	24	14		4
P. luteum series (non-ascosporic)	JQMD 396	Blanket	Hollandia, N.G.	6	53		9		3

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
P. luteum series (non-ascosporic) (cont.)	Fla A-24		Florida	3	102	106	97		1
P. luteum series (non-ascosporic)	Fla B-17	Canteen cover	Florida	3	77	34	18		4
P. luteum series (non-ascosporic)	USDA 1336.1			3	97	88	94		1
P. luteum series (non-ascosporic)	USDA 1336.1			3	96	98	90		1
P. luteum series (non-ascosporic)	Aust 41	Condenser label	New Guinea	3	91	32	18		4
P. luteum series (non-ascosporic)	Aust 82	Canvas	Horne Is.	3	50	25	15		4
P. majusculum Westl.	PQMD 50a	Rope	Hawaii	2	102	98	97		0
P. multicolor series	PQMD 45g	Shoe	Ledo, India	2	103	101	98		0
P. multicolor series	Fla B-40	Canvas	Florida	3	92	93	85		1
P. notatum Westling	Fla B-1	Tent	Florida	3	99	93	93		1
P. notatum Westling	Aust 40	Condenser label	New Guinea	3	105	102	96		0
P. piscarium Westling	PQMD 59a	Canvas	Finsch., N.G.	2	98	104	97		2
P. purpurogenum Stoll	PQMD 4n	Shoe	Bougainville Is.	1	105	92	83		2
P. purpurogenum Stoll	PQMD 4n	Shoe	Bougainville Is.	2	101	98	85		0
P. purpurogenum Stoll	PQMD 55d	Leather	Finsch., N.G.	2	103	96	97		1
P. purpurogenum series	PQMD 17j	Tent	Espiritu Santo Is.	1			105	100	1
P. purpurogenum series	PQMD 17j	Tent	Espiritu Santo Is.	8		105	102	105	0

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
P. roseo-purpureum Dieck (or near)	PQMD 8a	Cotton undershirt	Oro Bay, N.G.	2	95	103	103		0
P. rugulosum var. *atricolum* (Bain.) Thom	Fla B-16	Canteen cover	Florida	3	105	101	93		0
P. rugulosum var. *atricolum* (Bain.) Thom	Fla B-52	Tent	Florida	3	96	94	94		0
P. sanguineum Sopp.	Fla B-53	Tent	Florida	3	102	97	97		0
P. spinulosum Thom	PQMD 108b	Shoe	Florida	3	98	90	94		0
P. Steckii Zal.	PQMD 43a	Pliofilm	S.W. Pacific	2	106	101	108		0
P. Steckii Zal.	MIT 8	White cloth	Cambridge, Mass.	3	93	104	99		1
P. Steckii Zal.	MIT 8	White cloth	Cambridge, Mass.	3	96	87	91		1
P. sulfureum Sopp.	PQMD 52c	Shirt	Finsch., N.G.	2	107	104	112		0
P. Sweicickii Zal. (probably near)	PQMD 17h	Tent	Espiritu Santo Is.	1	103	98	91		1
P. Sweicickii Zal. (probably near)	PQMD 17h	Tent	Espiritu Santo Is.	8	106	100	101		1
P. tardum series	PQMD 4b	Shoe	Bougainville Is.	2		97	102		0
P. tardum series	PQMD 4b	Shoe	Bougainville Is.	7		104	103	99	0
P. tardum series	PQMD 24f	Netting	Russell Is.	1	103	101	103		0
P. tardum series	JQMD 236	Tarpaulin	Hollandia, N.G.	3	104	105	103		0
P. tardum series	JQMD 236	Tarpaulin	Hollandia, N.G.	6	100		102		0

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
P. virido-albus Sopp.	PQMD 53a	Leather	Finsch., N.G.	2	104	108	107		0
P. Westlingi Zal.	JQMD 238	Tarpaulin	Hollandia, N.G.	3	97	103	100		0
P. Westlingi Zal.	JQMD 238	Tarpaulin	Hollandia, N.G.	6	95		94		0
P. Westlingi Zal.	JQMD 311	Belt	Hollandia, N.G.	3	97	103	100		1
P. Westlingi Zal.	JQMD 311	Belt	Hollandia, N.G.	6	98		92		2
P. Wortmanni Klock.	PQMD 54b	Tent	Finsch., N.G.	6	97	102	96		1
Penicillium sp.	PQMD 4p	Tent	Finsch., N.G.	1		94	92	87	2
Penicillium sp.	PQMD 4p	Tent	Finsch., N.G.	7		100	101	100	1
Penicillium sp.	PQMD 17f	Tent	Espiritu Santo Is.	1			96	96	0
Penicillium sp.	PQMD 17f	Tent	Espiritu Santo Is.	8		100	104	101	0
Penicillium sp.	PQMD 17k	Tent	Espiritu Santo Is.	1			103	103	0
Penicillium sp.	PQMD 17k	Tent	Espiritu Santo Is.	8		107	99	101	1
Penicillium sp.	PQMD 26c	Canvas	Russell Is.	1	109	98	96		2
Penicillium sp.	PQMD 31b	Canvas	New Guinea	1	103	105	105		0
Penicillium sp.	PQMD 31e	Canvas	New Guinea	1	101	100	98		2
Penicillium sp.	PQMD 33b	Belt	New Guinea	1	100	100[1]	98		2
Penicillium sp.	PQMD 34a	Canvas	New Guinea	1	105	105[2]	96		2
Penicillium sp.	PQMD 47b	Wax paper	Ledo, India	2	94	94	85		1
Penicillium sp.	PQMD 49b	Web belt	Ledo, India	2	96	92	97		1
Penicillium sp.	PQMD 55a	Leather	Finsch., N.G.	2	100	108	102		1
Penicillium sp.	PQMD 57b	Imitation leather	Finsch., N.G.	2	101	98	107		0
Penicillium sp.	PQMD 58a	Leatherette	Finsch., N.G.	2	108	99	103		1
Penicillium sp.	PQMD 58f	Leatherette	Finsch., N.G.	2	104	105	97		1

[1] Figure 2*D*. [2] Figure 2*F*.

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Penicillium sp. (cont.)	PQMD 62b	Corncob pipe	Finsch., N.G.	2	104	108	102		0
Penicillium sp.	PQMD 64d	Shirt	Finsch., N.G.	2	97	99	96		1
Penicillium sp.	PQMD 65a	Canvas	Finsch., N.G.	2	93	101	94		1
Penicillium sp.	PQMD 71a	Canvas glove	Finsch., N.G.	3	92	96	99		0
Penicillium sp.	PQMD 71b	Canvas glove	Finsch., N.G.	3	97	102	95		0
Penicillium sp.	PQMD 72b	Leather	Finsch., N.G.	3	95	101	95		0
Penicillium sp.	PQMD 72b	Leather	Finsch., N.G.	3	92	97	89		0
Penicillium sp.	PQMD 111b	Rope	Florida	3	87	66	34		4
Penicillium sp.	PQMD 124b	Duck	Panama	3	100	105	103		0
Penicillium sp.	JQMD 179	Cork gasket	Jeffersonville, Ind.	3	107	105			0
Penicillium sp.	Fla A-21			1		101	94	111	1
Penicillium sp.	Fla B-25	Cloth	Florida	3	100	93	100		0
Penicillium sp.	Fla B-39	Canvas	Florida	3	91	90	88		2
Penicillium sp.	Fla B-41	Canvas	Florida	3	84	101	100		0
Penicillium sp.	Fla B-64	Legging	Florida	3	94	98	96		0
Penicillium sp.	Fla B-54	Tent	Florida	3	82	46	27		4
Penicillium sp.	Fla B-65	Legging	Florida	3	100	96	99		0
Penicillium sp.	Fla C-5	Web belt	Florida	3	105	108	110		0
Penicillium sp.	Aust 114	Blood serum	Sydney, Aust.	3	107	105	103		1
Pestalotia sp.	Aust 8	Canvas	New Guinea	3	75	57	56		3
Pestalotia sp.	PQMD 2d	Duck	Bougainville Is.	1		32	10	0	4
Pestalotia sp.	PQMD 2d	Duck	Bougainville Is.	7		31	7	4	4
Pestalotia sp.	PQMD 119a	Canvas	Panama	3	85	75	45		2
Pestalotia sp.	PQMD 119b	Canvas	Panama	3	80	70	56		3
Pestalotia sp.	PQMD 119b	Canvas	Panama	8		38	17	9	4
Pestalotia sp.	PQMD 120b	Tarpaulin	Panama	3	79	69	65		1

Pestalotia sp.	PQMD 119b	Canvas	Panama	8		38	17	9	4
Pestalotia sp.	PQMD 120b	Tarpaulin	Panama	3	79	69	65		1

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Pestalotia sp. (cont.)	PQMD 121b	Tarpaulin	Panama	3	104	100	103		2
Pestalotia sp.	PQMD 121L	Tarpaulin	Panama	3	86	80	70		3
Pestalotia sp.	PQMD 121L	Tarpaulin	Panama	8		81	50	37	4
Pestalotia sp.	PQMD 125a	Duck	Panama	3	84	84	82		1
Pestalotia sp.	Fla C-72	Cord	Florida	3	86	70	67		1
Phoma sp.	PQMD 13e	Canvas	Espiritu Santo Is.	1	77	65	67		1
Phoma sp.	PQMD 48c	Tent	Ledo, India	2	63	54	35		2
Phoma sp.	PQMD 120k	Tarpaulin	Panama	3	82	67	39		3
Phoma sp.	PQMD 120k	Tarpaulin	Panama	8	63	31	9		3
Phoma sp.	PQMD 122f	Duck	Panama	3	75	64	54		1
Phoma sp.	PQMD 121h	Tarpaulin	Panama	3	64	30	20		3
Phoma sp.	Fla B-30	Cloth	Florida	3	52	26	18		2
Pullularia pullulans (D By.) Berkh.	PQMD 72h	Leather	Finsch., N.G.	3	96	94	97		0
Pullularia pullulans (D By.) Berkh.	PQMD 72h	Leather	Finsch., N.G.	3	92	98			0
Pullularia pullulans (D By.) Berkh.	PQMD 72h	Leather	Finsch., N.G.	8	100	98	104		1
Pullularia pullulans (D By.) Berkh.	PQMD 122b	Duck	Finsch., N.G.	3	95	95	98		0
Pullularia pullulans (D By.) Berkh.	JQMD 364	Belt	Hollandia, N.G.	3	102	98	90		0
Pullularia pullulans (D By.) Berkh.	Fla B-37		Florida	3		107	106		0
Pullularia pullulans (D By.) Berkh.	Fla F-44		Florida	3	103	105	93		0

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Pyrenochaeta sp.	PQMD 29b	Tarpaulin	Kauai, T.H.	2	51	27	22		4
Rhizopus arrhizus Fisch.	PQMD 46c	Rope	Ledo, India	2	101	103	103		0
Sarcopodium fuscum (Corda) Sacc.	PQMD 28a	Tent	Kauai, T.H.	1	46	32	14		4
Sarcopodium fuscum (Corda) Sacc.	PQMD 28a	Tent	Kauai, T.H.	3	25	11	5		0
Scopulariopsis brevicaulis (Sacc.) Bain.	Fla C-28	String	Florida	3	100	103	92		1
Septomyxa affinis (Sherb.) Woll.	PQMD 40b	Tent	Karachi, India	2	106	101	87		0
Sphaeropsis sp.	PQMD 46h	Rope	Ledo, India	2	103	95	93		2
Sphaeropsis sp.	PQMD 46h	Rope	Ledo, India	3	98	101	96		2
Sporotrichum sp.	PQMD 4f	Shoe	Bougainville Is.	1	100	92	89		0
Stachybotrys atra Corda	Fla B-10	Tent	Florida	3	18	19	19		4
Stachybotrys atra Corda	Fla B-31	Cloth	Florida	3	23	20	16		4
Stachybotrys atra Corda	Fla B-62	Tent	Florida	3	56	41	31		2
Stachybotrys sp.	Fla C-10	Shower curtain	Florida	3	0				4
Stemphylium consortiale (Thüm) Gr. & Sk.	PQMD 41b	Rope	Karachi, India	2	66	48	32		4

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Syncephalastrum racemosum (Cohn) Schr.	PQMD 57a	Imitation leather	Finsch., N.G.	2	106	102	102		1
Syncephalastrum racemosum (Cohn) Schr.	Fla B-23	Canteen cover	Florida	3	98	97	99		1
Syncephalastrum racemosum (Cohn) Schr.	Fla B-35	Cloth	Florida	3	95	96	100		1
Thielavia Sepedonium Emm.	PQMD 46a	Rope	Ledo, India	2	32	16	0		4
Thielavia Sepedonium Emm.	PQMD 47g	Wax paper	Ledo, India	2	15	0			4
Trichoderma viride (Pers.) Harz	PQMD 6a	Tent	Bougainville Is.	1	18	10	8		4
Trichoderma viride (Pers.) Harz	PQMD 66a	Strap	Finsch., N.G.	2	29	18	7		4
Trichoderma viride (Pers.) Harz	PQMD 13b	Canvas	Espiritu Santo Is.	1		91	101	98	0
Trichoderma viride (Pers.) Harz	PQMD 18a	Tent	Espiritu Santo Is.	1	72	36	13		2
Trichoderma viride (Pers.) Harz	PQMD 18a	Tent	Espiritu Santo Is.	3		78	71	71	4
Trichoderma viride (Pers.) Harz	PQMD 18e	Tent	Espiritu Santo Is.	1	47	19	15		4
Trichoderma viride (Pers.) Harz	PQMD 21b	Tarpaulin	Espiritu Santo Is.	1	60	31	16		4
Trichoderma viride (Pers.) Harz	PQMD 22d	Tarpaulin	Russell Is.	1	36	25	16		4
Trichoderma viride (Pers.) Harz	PQMD 22e	Canvas	Russell Is.	1	53	33	18		3

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experimen
					6 Days	9 Days	12 Days	15 Days	
Trichoderma viride (Pers.) Harz (cont.)	PQMD 33a	Canvas	New Guinea	1	91	72[1]	58		4
Trichoderma viride (Pers.) Harz	PQMD 46d	Rope	Ledo, India	2	56	29	16		4
Trichoderma viride (Pers.) Harz	PQMD 63d	Cotton rope	Finsch., N.G.	2	100	99	98		0
Trichoderma viride (Pers.) Harz	PQMD 120o	Tarpaulin	Panama	3	98	94	44		3
Trichoderma viride (Pers.) Harz	JQMD 314	Belt	Hollandia, N.G.	3	86	65	31		3
Trichoderma viride (Pers.) Harz	JQMD 315	Belt	Hollandia, N.G.	3	56	26	28		3
Trichoderma viride (Pers.) Harz	JQMD 474	Raincoat	Hollandia, N.G.	3	67	27	20		4
Trichoderma viride (Pers.) Harz	Fla A-4		Florida	1	47	19	15		4
Trichoderma viride (Pers.) Harz	Fla B-3	Tent	Florida	3	101	55	22		4
Trichoderma viride (Pers.) Harz	Fla C-55	Socks	Florida	3	31	16	7		4
Trichophyton sp.?	PQMD 199	Wool fabric	Philadelphia	8	105		101	104	1
Trichophyton sp.?	PQMD 208	Wool fabric	Philadelphia	8	106		99	106	1
Tritirachium dependens Limb. & *Asperg. versicolor* (Vuill.) Tir.	PQMD 4g	Shoe	Bougainville Is.	1	105	101	101		1

[1] Figure 2*C*.

[1] Figure 2C.

TABLE II—*continued*

Species	Culture Number	Source	Locality	Method Variant	Per Cent Strength Retained				Growth at End of Experiment
					6 Days	9 Days	12 Days	15 Days	
Tritirachium sp.	PQMD 164	Veg. ivory buttons	Tropics	8	103		100	101	1
Verticillium sp.?	Fla B-51	Canvas	Florida	3	54	32	27		3
Verticillium sp.?	Fla B-59	Tent	Florida	3	51	25	15		2
Verticillium sp.?	Fla C-7	Web belt	Florida	3	70	41	29		0
Verticillium sp.?	PQMD 125e	Duck	Panama	3	27	23	16		1
Zygorhynchus Moelleri Vuill. with internal parasite	Fla A-29		Florida	3	36	14	7		2
Undet. Dematiaceae	PQMD 14c	Rope	Espiritu Santo Is.	1	94	103	100		0
Undet. Dematiaceae	PQMD 14c	Rope	Espiritu Santo Is.	7		101	103	99	1
Undet. Dematiaceae	PQMD 69c	Coat	Finsch., N.G.	3	96	94	100		1
Undet. Moniliaceae	PQMD 119c	Canvas	Panama	3	91	71	62		2

The figures cited in the right hand column to indicate the estimated amount of growth at the end of the incubation period are as follows: (0) meaning no growth visible to the naked eye (FIG. 2 *A, K*); (1) a small amount of growth in the form of a scarcely visible fuzziness which might be expected simply as a result of spore germination and the utilization of a small amount of food material available as impurities (FIG. 2*D*); (2) slightly more growth, often accompanied by sparse or inhibited fruiting and not necessarily an indication of cellulose utilization (FIG. 2*F, G*); (3) well developed fungus growth which may always be taken as indicative of cellulolytic activity (FIG. 2*C*); and (4) meaning that the fabric is covered or nearly so with a well developed growth typically indicative of strong activity (FIG. 1; 2*B, E, H, I, J, L;* 3).

SUMMARY AND COMMENT

Results are reported of a series of assays to determine roughly the cellulolytic activity, as measured by decline in tensile strength of cotton fabric, of certain fungi isolated from textiles and related materials deteriorated in the tropics. The total number of assays was 584. These represent 453 cultures distributed among 45 genera. Of the 453 cultures, 310 are identified to series, species or variety and 143 are determined only to genus or section of genus. The total of 310 cultures determined beyond generic rank, *i.e.*, the number of species or approximate species, is 91. The great majority are the imperfect stages of Ascomycetes, *i.e.*, Fungi imperfecti; a few are perithecial and a few are Mucorales. No Basidiomycetes are included. Cellulolytic activity is indicated for 41 of the determined species whereas 50 gave negative results.

The following summarizing comments are offered:

1. *Mucorales.* Six species were tested: *Absidia capillata, Cunninghamella echinulata, Lichthemia Regnieri, Mucor fumosus, Rhizopus arrhizus,* and *Syncephalastrum racemosum.* In all cases the results were negative. A seventh culture, *Zygorhynchus Moelleri* (Fla *A–29*), presented an interesting case. Continued investigation of its apparent cellulolytic activity finally demonstrated this to be due to the action of an internal Dematiaceous parasite, which, following inoculation of the cloth, grew out of its host and

attacked the fabric. A few Mucorales have been tested previously (**1, 2, 3, 9, 13, 16**), and the results always have been negative. Evidence is accumulating that the Mucorales lack the ability to utilize cellulose as a source of carbon or to degrade it directly.

2. *Aspergillus.* A total of 125 cultures were tested. These represent all the 14 groups of Thom and Raper (**15**) except the *A. clavatus* and *A. candida* groups. Of the remaining 12 groups, cellulolytic activity was demonstrated in five. The results are summarized briefly below by group.

a. The A. glaucus group. Five cultures representing *A. Chevalieri* and its variety *intermedius* and four of *A. repens* were assayed. The results were in all cases clearly negative. This is in agreement with results obtained by Galloway (**2**), although such forms are sometimes recorded (**2, 13**) as occurring on cotton goods.

b. The A. fumigatus group. The one culture of *A. Fisheri* tested gave positive results in each of three assays, whereas three strains of *A. fumigatus* were positive and three negative. The results obtained here, together with those recorded previously (**6, 9, 13, 16**), indicate that capacity to decompose cellulose probably is general in the group.

c. The A. nidulans group. Seven strains of *A. unguis* gave clearly negative results. The one culture of *A. nidulans* tested gave doubtful results in one case and negative in a somewhat modified repeat assay. Previous tests (**2, 9, 13**) of what has been recorded as *A. nidulans* have given contradictory results.

d. The A. ustus group. Cultures of *A. ustus* and its variety *laevis* behaved rather peculiarly in that they made considerable growth without causing a pronounced decline in tensile strength. It appears possible, or even probable, that cellulolytic ability will be found to be general in the group under a relatively restricted optimal set of conditions.

e. The A. flavipes group. Weak cellulolytic activity was demonstrated for all three of the cultures of *A. flavipes* tested. This is in conformity with the results of later more detailed work (unpublished) in connection with the present program. Galloway (**2**) recorded its action as strong based on visual growth on filter paper.

f. The A. versicolor group. The numerous cultures of *A. versicolor* and *A. Sydowi* were all negative and give strong indication that members of the group are unable to attack cellulose despite previous literature records (**2, 9**) to the contrary.

g. The A. terreus group. The seven cultures of *A. terreus* which were tested grew and fruited readily and consistently on test strips. This perhaps is the most strongly and consistently cellulolytic species of the genus. Other workers (**2, 9**) have obtained similar results.

h. The A. niger group. This has been a very controversial group in deterioration laboratories with several early literature records (**13**) claiming cellulolytic properties for its members, partly on circumstantial evidence. Of the numerous cultures here tested, positive results were obtained for only one, and possibly a second, both of which were later determined as *A. luchuensis.* No effort has been made to identify the other cultures beyond the group category, except to ascertain that there were no additional cultures of *A. luchuensis* among them. A more detailed study of cellulolytic activity in the *A. niger* group is in progress.

i. A. Wentii group. The single culture tested gave negative results on both bleached sheeting and gray duck. Others (**2, 13**) have indicated positive results.

j. The A. Tamarii group. No activity could be demonstrated in three isolates determined as *A. Tamarii.* Galloway (**2**) recorded moderate growth on filter paper and Furry & Zametkin (**1**) claimed decline in strength of cotton duck.

k. The A. flavus-Oryzae group. No cellulolytic activity was demonstrated for the numerous cultures identified as *A. flavus* and *A. Oryzae.* This conflicts with results obtained by others (**1, 2, 13**).

l. The A. ochraceus group. Results were negative for the two cultures identified as *A. ochraceus.* Galloway (**2**) reported moderate visual growth on filter paper.

3. *Penicillium.* A total of 126 assays were conducted, including 94 cultures of which 62 are determined approximately to species and 32 remain unidentified. The only clearly positive results obtained were for certain, or most, of the 9 strains of *P. luteum*

that were tested, for some strains of the *P. citrinum* series and for two unidentified forms, *i.e.*, PQMD *111b* and Fla *B–54*. *P. citrinum* is isolated extremely commonly from fabrics and similar materials exposed in the tropics and should be given further attention. In the review of Thaysen and Bunker (**13**) of twenty years ago 19 species of *Penicillium* were recorded as having given positive results based mostly on visual growth on filter paper or on the clearing of a cellulose agar medium. Of the nineteen species recorded there as cellulolytic, six appear in the table of this paper. The results correlate for only one species, *P. luteum*. The positive results previously recorded for *P. chrysogenum, P. notatum, P. purpurogenum, P. rugulosum,* and *P. spinulosum* could not be confirmed in the present study. Taxonomic confusion in such a difficult group could account for this, at least in part. Since the appearance of the book by Thaysen and Bunker only scanty mention has been made of members of the genus relative to cellulose decomposition (**1, 2, 3, 9**).

4. *Moniliaceae.*

a. Fusarium. The results of tests of 34 isolates representing 10 species and varieties indicate that substantial cellulolytic activity probably is the rule in this genus. This is in agreement with the several scattered literature records (**1, 2, 9, 13**) of tests of undetermined species. The results indicated in the accompanying table are not to be considered significant to the extent that they may be considered as representing species differences with respect to cellulolytic ability.

b. Gliocladium. The eight cultures listed under *Gliocladium* sp. represent a single species. It is the species represented by Weindling's culture G–1, which, together with one or two other strains, has been used in the production of gliotoxin and viridin. It has generally been referred to as *Gliocladium fimbriatum* or *Trichoderma viride,* but is distinct from either of these. This question is being taken up in another paper. Cellulolytic activity appeared to be strong in some strains and weak or absent in others and some evidence of instability in this respect has been noted. One strain of *G. roseum* was tested and proved to be cellulolytic.

c. Humicola. The 7 cultures tested, representing 2 species, all were found to be strongly cellulolytic. They are considered un-

identifiable, pending the appearance of a better basic taxonomy of the genus.

d. Trichoderma viride. Of seventeen strains tested, all proved to be relatively strongly cellulolytic. The species is isolated very frequently from fabrics and is of common and universal occurrence throughout the tropic and temperate parts of the world. It is fairly generally known as a cellulose destroyer, and is considered to be active in the soil in this respect.

e. Tritirachium. T. dependens and an undetermined species were clearly negative.

f. Other Moniliaceae. Results for members of several other less prominent genera for which space cannot be taken for detailed summarization, may be found by turning the pages of the table. Genera represented are: *Acladium, Acremonium, Cephalosporium, Microsporum, Oospora, Paecilomyces, Sarcopodium, Scopulariopsis, Septomyxa, Sporotrichum,* and *Verticillium.* Occasional mention of members of some of these genera will be found in papers listed in the bibliography. Some are in genera where specific identification is impossible, and even where the possibility of a really meaningful generic disposition is extremely doubtful.

5. *Dematiaceae.*

a. Alternaria. Five cultures were tested, all of which were identified with strains commonly referred to *A. tenuis.* Relatively uniform and fairly strong activity is indicated. Previously recorded tests of unidentified species (**4, 9, 13**) have indicated similar results.

b. Brachysporium. Six cultures were tested without any attempt having been made to match them or to identify any of them to species. Activity similar to that of the more or less related *Alternaria* group is indicated.

c. Cladosporium herbarum. Of universal occurrence on fabrics and other materials exposed in nature, it is in need of further attention both taxonomically and physiologically. The results for the many cultures tested here and from various literature records (**4, 9, 13**) indicate varying, and, on the whole, not strong cellulolytic activity for the forms commonly loosely placed here.

d. Curvularia. Ten isolates distributed among three species gave results similar to those obtained for *Alternaria* and *Brachy-*

sporium. Within the accuracy of these tests no cellulolytic differences are noted among the species.

e. Gliomastix convoluta. Eight cultures tested were all strongly cellulolytic. Culture *4c* was selected for detailed nutritional studies in a Quartermaster sponsored project, but the results are not as yet published. The identification is uncertain.

f. Memnoniella echinata. All of a total of eleven cultures tested were strongly cellulolytic.

The figures in the table are interesting in several respects. Since little mycelial growth is made on a cellulose substratum the visual growth rating refers largely to the amount of fruiting. In all instances the fabric was either covered at the end of twelve days with black sporulation or there was no fruiting at all. Intermediates did not occur. The contrast among strains in this respect was more striking than among strains of any other species assayed. Cellulolytic activity appeared to be correlated positively with fruiting even though it was fairly strong where no fruiting occurred. Interpreting the table in the light of the work of Marsh & Bollenbacher (**7**), who demonstrated the need for an external source of biotin in all of six cultures of the species which they tested, one might postulate that both biotin requiring and non-biotin requiring strains exist. Strain Aust *86* in the table was among those shown (l.c.) to require biotin. PQMD 1c would probably be supplied a sufficient amount by the test method used. Under the more highly purified method variants 2 and 3, by which the other strains were tested, biotin should be absent or nearly so.

6. *Tubercullariaceae*. Represented in these tests by three species of *Myrothecium* of which *M. verrucaria* (Syn.: *Metarrhizium glutinosum* Pope) and the very closely allied *M. roridum* are among the strongest cellulose decomposers yet found. (Cf. also **4**, **10**, **18**.) A culture of *M. inundatum* exhibited weak or negative action. Detailed nutritional studies are in progress as a part of the present program.

7. *Stilbaceae*. Represented only by a culture identified as *Arthrosporium* sp., which evidenced no activity.

8. *Sphaeropsidales*.

a. Botryodiplodia Theobromae. The results obtained for three cultures which were tested indicated negative, or at most, very weak activity. This is a common tropical species capable of invading the living tissues of a wide variety of plants. It is isolated from fabrics very frequently but there is as yet no evidence that it is there other than as spores deposited from the atmosphere. It should be given further attention.

b. Coniothyrium, Phoma, and *Pyrenochaeta.* A complex of pycnidial forms having very small ellipsoid spores and referable to these genera are found producing pycnidia on fabrics with considerable frequency, often accompanied by staining. Numerous cultures are at hand and those tested proved to be strongly cellulolytic.

9. *Ascomycetes*

a. Chaetomium. Fifteen cultures were tested. These included seven of *C. funicolum,* six of *C. globosum,* and two of *C. indicum.* Since *C. globosum* has long been known as a strong cellulose-destroyer (**11, 13, 14**) and since a wide range of additional species were tested recently (**5**), the results presented here are not of great interest. They do, however, suggest that within limits there is no great variation in different strains of the three species, and that a given strain of any one of the three species is likely to be about as strong cellulolytically as the standard strain, USDA *1042.4,* used in assessment and procurement testing of cotton fabrics.

b. Thielavia Sepedonium. The two cultures tested exhibited action of the same order of magnitude as that of species of *Chaetomium.*

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EXPLANATION OF FIGURES

FIG. 1. Cloth test strips incubated nine days under method variant 3 of Table I. (Magnification × 1.) At left uninoculated control, at right inoculated with *Curvularia brachyspora*, Fla *B–8*.

FIG. 2. Cloth test strips inoculated with various fungi and incubated nine days under method variant 1 of Table I. (Magnification × 1.) *A*, Control, incubated without having been inoculated; *B, Chaetomium globosum*, PQMD *32b; C, Trichoderma viride*, PQMD *33a; D, Penicillium* sp., PQMD *33b; E, Chaetomium funicolum*, PQMD *33c; F, Penicillium* sp., PQMD *34a; G, Aspergillus niger*, PQMD *34c; H, Chaetomium funicolum*, PQMD *34d; I, Humicola* sp., PQMD *34e; J, Myrothecium verrucaria*, PQMD *34f; K, Cunninghamella echinulata*, PQMD *35c; L, Chaetomium globosum*, USDA *1042.4*. The spotting of strips *A, D, F*, and *K* is due to uneven contact with the plate glass on which they were placed for photographing immediately after removal from the incubation vessels; little or no fungous growth is visible in the photograph.

FIG. 3. Strips of gray cotton duck incubated twelve days under method variant 5 of Table I. (Magnification × 1.) At left uninoculated control, at right inoculated with *Aspergillus luchuensis*.

THREE ZOÖPAGACEAE THAT SUBSIST BY CAPTURING SOIL AMOEBAE

CHARLES DRECHSLER [1]

(WITH 4 FIGURES)

As the unusually cool humid weather that prevailed during the summer of 1946 in Maryland, Virginia, and North Carolina furnished conditions of temperature and moisture under which the fungi destructive to eelworms and rhizopods have been found developing most abundantly in the laboratory, it was expected that decaying plant materials collected in later months might yield many zoöpagaceous forms not ordinarily encountered. However, when vegetable detritus taken from woods, fields, and gardens, in various localities within the states mentioned, was planted on Petri plates of maizemeal agar suitably permeated with *Pythium* mycelium, the resulting cultures were in most instances ruined through an overwhelming development of plasmodia belonging to different species of Myxomycetes. Usually these plasmodia suppressed multiplication of rhizopods and eelworms at an early stage, and thereby forestalled virtually all development of fungi habitually subsisting on these animals. In other instances zoöpagaceous fungi became established and began to sporulate in noticeable quantity, but would then be obliterated under shifting streams of slime. In a few cultures, fortunately, plasmodia failed to develop; and here abundant multiplication of rhizopods and eelworms took place, together with development of fungi destructive to them. Among these fungi were observed the three predaceous forms herein described as new members of the Zoöpagaceae.

Trouble from plasmodia of Myxomycetes has been experienced now and then in earlier years, though not on a serious scale. Or-

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dinarily cultures prepared with decaying plant materials collected outdoors in Maryland and Virginia are less likely to be ruined by slime molds than from infestation with mites, annelids, tardigrades, or rotifers. Mites have been found especially troublesome in cultures planted with decaying material taken from greenhouses, or with decaying plant detritus collected outdoors in such warm states as Florida and Louisiana. While these pests do not necessarily prevent vegetative development of nematode-destroying and rhizopod-destroying fungi deep under the surface of an agar culture, they usually grind up the surface layer so badly by their manner of feeding that the reproductive apparatus of all except the most robust Zoöpagaceae is broken up beyond all hope of recognition, and often the conidial apparatus of the sturdiest forms is mangled so severely as to render it unfit for study. The use of herbarium poisons, such as naphthalene and paradichlorobenzene, in mite-infested cultures has not been found advantageous since rhizopods and nematodes succumb quickly in the presence of these volatile compounds. Storage of the cultures at low temperatures similarly entails some disadvantage, for thereby not only the infesting mites but also the microscopic animals and associated fungi similarly preferring warm conditions—the very forms, indeed, that could be expected to be most characteristic of mite-infested material—are largely if not wholly suppressed.

The minute annelids that frequently appear in Petri plate cultures five to ten days after decaying material has been added cause even more serious damage than mites, since they burrow through the agar at all depths, creating havoc in their paths. These unwanted animals seem to hatch most quickly when a rather soft agar medium is employed, or when some free liquid water collects around the material planted, yet may become troublesome even when firm agar is employed and when little moisture condenses. Difficulty with them may be largely obviated, as a rule, by taking care in collecting leaf mold or other decaying plant detritus to include no considerable admixture of sand, clay, loam, or gravel from the underlying soil. Destruction by annelids is exceedingly frequent in cultures planted with ordinary soil from gardens and fields. Despite the manifestly thoroughgoing distribution of the Zoöpagaceae on all arable land, the difficulties attending annelid

infestation have strongly discouraged the use of firm soil in preparing cultures intended for the study of these fungi.

ANOTHER ACAULOPAGE WITH LONG SLENDER ROD-LIKE CONIDIA

Several maizemeal-agar plate cultures that when completely overgrown with mycelium of *Pythium ultimum* Trow had been further planted through addition of decaying detritus taken on August 20, 1946, from an old pile of weeds in a field near Mayo, Maryland, were found after twenty-seven days to contain a sparse branching mycelium composed of narrow colorless aseptate hyphae (FIG. 1, *A–G*) to which were attached many individual *Amoebae* generally similar to one another. The captives commonly measured from 20 to 40 μ in diameter when drawn into a moderately rounded shape. They were surrounded individually by a thin yet distinctly visible pellicle, which, except in regions where pseudopodia were being extended, was handsomely disposed in delicately rippled folds. Their protoplasm, though colorless and for the most part of only moderately densely granular texture, was reduced in transparency by numerous vacuole-like inclusions of globular or ellipsoidal shape. These inclusions were mostly less than 5 μ wide, but in some animals (FIG. 1, *C, D, E*) one or two among them measured about 10 μ in their greatest dimension and thus rivaled the contractile vacuole (FIG. 1, *A–G: v*) in size. Some animals contained, besides, one or more commonplace digestive vacuoles (FIG. 1, *A, E*). While the single nucleus (FIG. 1, *A–G: n*) appeared to be generally of prolate ellipsoidal shape, the difference between its length and its width was often relatively small. In comparison with the size of the animal the nucleus seemed unusually large, its length varying commonly from 8 to 12.5 μ, and its width from 7 to 11 μ. It showed, distributed in peripheral positions, some 35 to 50 globose or slightly flattened bodies about 1 μ or slightly less in width. Among the *Amoebae* previously reported as being destroyed by members of the Zoöpagaceae the animal here concerned seemed to resemble most the species found habitually captured by my *Stylopage rhabdospora* (**5**: 374–377) and my *S. cephalote* (**7**: 144–148). However the obscurely globuliferous structure of the protoplasm, together with

the apparently larger number and smaller size of the chromatin bodies, gives reason for presuming that the animal is specifically distinct not only from the one earlier observed serving as prey for the two species of *Stylopage*, but also from the one found parasitized by my *Cochlonema euryblastum* (**10**: 283–289). In the *Amoeba* parasitized by *C. euryblastum*, besides, the nucleus appeared appreciably smaller relative to the entire animal than in the *Amoeba* of globuliferous protoplasmic texture.

Following narrow perforation of its integument each captured *Amoeba* was soon invaded by a haustorium (FIG. 1, *A–E*), or at times by two haustoria (FIG. 1, *F, a, b; G, a, b*). As in *Stylopage rhabdospora* the absorptive organ here was of the familiar pedicellate type. Ramification of the assimilative elements was mainly dichotomous, though irregularity of branching appeared rather often, especially in the more elaborately developed haustoria. The number of terminal branches occasionally exceeded sixteen (FIG. 1, *F, b; G, b*). The assimilative elements appeared generally to be of about the same width as mycelial hyphae. Owing to the globuliferous character of the animal's protoplasm the haustorium was only somewhat indistinctly visible during the earlier stage of its development, but after most of the protoplasm had been assimilated it emerged more clearly into view (FIG. 1, *H, I*). Later, when the *Amoeba* was depleted of its digestible substance, the contents of the haustorium were withdrawn backward into the parent hypha; the empty tubular envelope thereupon soon fading from sight together with the empty pellicle loosely surrounding it.

In one of the plate cultures an area occupied by mycelium of the fungus showed a few captured *Amoebae* (FIG. 1, *J, K*) which, though of about the same size as those taken in large numbers, were distinguished by protoplasm of commonplace granular texture and by a smaller nucleus (FIG. 1, *J, n; K, n*) that contained a single central endosome instead of many peripheral bodies. Despite careful scrutiny it was not possible to trace any mycelial connection between the hyphae holding fast these few animals of a different species and the hyphae holding fast the much more numerous captives. The fungus represented in these latter hyphae, therefore, cannot yet be reckoned among the small number of zoö-

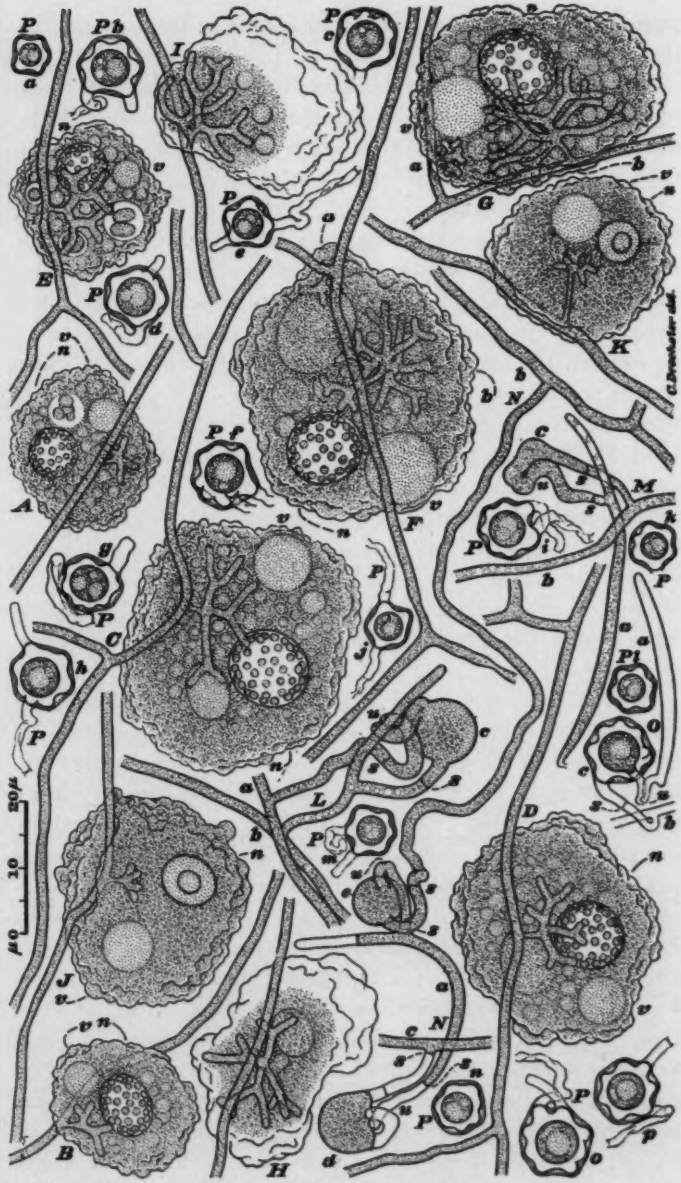

FIG. 1. *Acaulopage baculispora.*

pagaceous forms definitely known to subsist on two or more distinct species of rhizopods.

The fungus gave rise abundantly to both asexual and sexual reproductive apparatus. In initiating asexual reproduction a prostrate mycelial filament would at mostly rather short intervals put forth erect narrow processes that in their upward growth widened out until they had attained a height of about 15 μ (FIG. 2, *A, a*). Thereupon they continued elongating either at a uniform width, or at a slightly diminishing width, until on attaining a definitive length of 25 to 65 μ (FIG. 2, *A, b*) a cross-wall was formed a few microns above the base to delimit, in each instance, a rod-shaped or somewhat filamentous erect conidium from the short, often slightly tapering sterigma below (FIG. 2, *A, c–g; B, a–e*). Collectively the conidia in an undisturbed sporulating tract offered a handsome bristling display. They readily became disjointed, however, when they were jostled by roving eelworms or rotifers. The procumbent mycelial hyphae, including those beset with numerous denuded sterigmata (FIG. 2, *C, a–e*), came thereby to be partly obscured amid a disorderly confusion of detached spores (FIG. 2, *D, a–z; E, a–w*). Many of the spores soon germinated by putting forth a germ tube somewhat obliquely from the basal or the distal end (FIG. 2, *F*).

Although in some measure sexual reproduction took place rather early through union of gametangia borne on branches contributed from two mycelial hyphae (FIG. 1, *L, a, b*), it proceeded far more briskly after the surface of the substratum was liberally strewn with detached conidia. In the species here concerned, as in many related forms, most units of sexual apparatus originated from pairs of gametangia whereof one came from a germinating conidium (FIG. 1, *M, a; N, a; O, a.* FIG. 2, *Ga, a; H, a; I, a*) while the other came from a mycelial filament (FIG. 1, *M, b; N, b, c; O, b.* FIG. 2, *G, b; H, b; I, b*). The paired gametangia appeared to become delimited by basal cross-walls (FIG. 1, *L, s; M, s; N, s; O, s.* FIG. 2, *G, s; H, s; I, s*) at about the same time they began to fuse at their tips. Sometimes a gametangium contributed from the mycelium was found borne on a branch so short as to appear virtually sessile on the mycelial hypha (FIG. 1, *N, c.* FIG. 2, *G, b; H, b*); frequently it was borne on a branch 5 to 25 μ in length

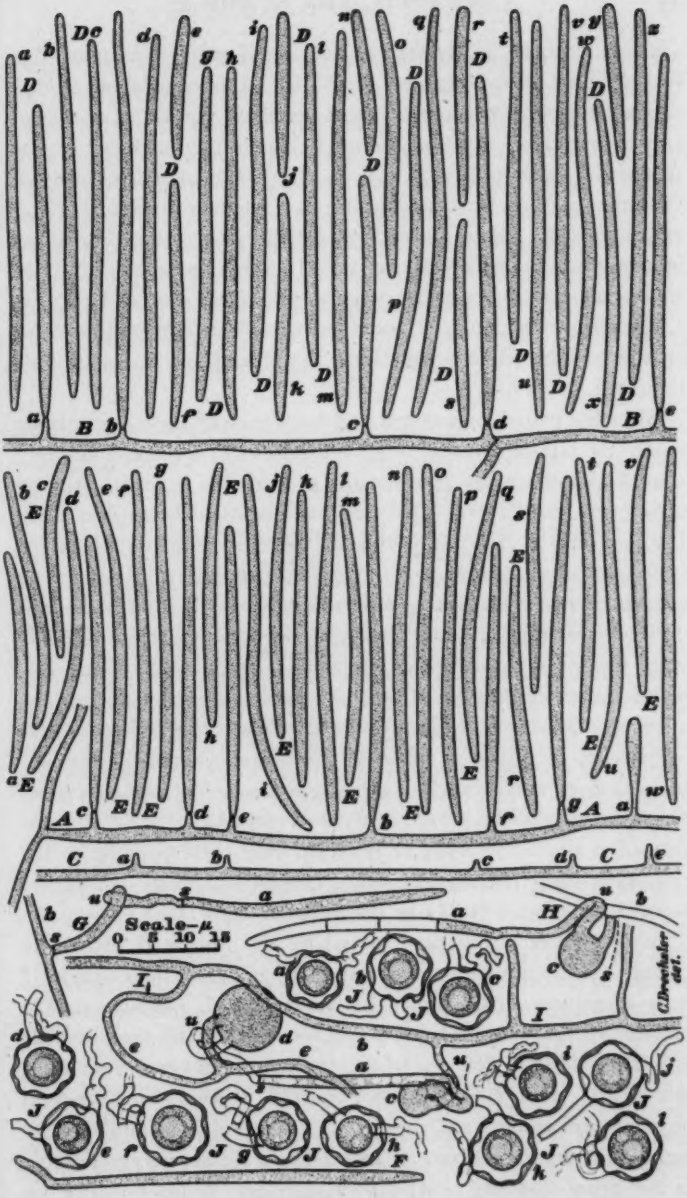

FIG. 2. *Acaulopage baculispora.*

(FIG. 1, *L, a, b; M, b; O, b*); and occasionally it terminated a branch about 100 μ long (FIG. 1, *N, b*). Gametangia of conidial origin often were borne on a short proximal portion of the germ hypha (FIG. 1, *M, a*), but frequently, again, they included a portion of the spore itself (FIG. 2, *G, a; I, a*), and sometimes, indeed, the entire spore seemed to function as a sexual cell (FIG. 1, *O, a.* FIG. 2, *H, a*). Often a conidium collaborated in the production of two units of sexual apparatus. In these instances it would sometimes extend a zygophoric germ tube from each end (FIG. 2, *I, a*), but at other times it would extend one such germ tube from an end position while putting forth the other from a median position (FIG. 1, *N, a*).

After the paired gametangia had fused, one of them—very commonly the one of mycelial origin—would form a globose swelling (FIG. 1, *M, c*) usually at a distance of 5 to 10 μ from the place of union (FIG. 1, *M, u*). This swelling continued to expand (FIG. 1, *L, c.* FIG. 2, *H, c; I, c, d*) until the remaining portion of the fusion cell was empty of protoplasmic material. The spherical zygosporangium thus brought into being (FIG. 1, *N, d*) then underwent the usual internal changes whereby its cellular contents were converted into a zygospore with a thick, boldly verrucose, distinctly yellowish wall. In mature units of sexual apparatus the zygospore, loosely enveloped by the somewhat collapsed sporangial membrane, contained a spherical protoplast wherein a granular parietal layer surrounded a central reserve globule, or sometimes two reserve globules (FIG. 1, *P, a–p.* FIG. 2, *J, a–l*). Often, besides, a smaller lustrous body, apparently corresponding to the refringent body present in mature oöspores of many oömycetes, seemed indistinctly visible in the granular layer.

In the genus *Acaulopage*, to which the fungus is manifestly referable, it invites comparison especially with the three species I have described earlier under the binomials *A. macrospora* (2: 189–191), *A. stenospora* (9: 256–258), and *A. ischnospora* (12: 263–269), all of which give rise to narrow erect conidia on short sterigmata. It is clearly distinct from *A. ischnospora* by reason of its shorter conidia and their lack of an empty membranous appendage at the tip. From *A. stenospora* it differs very noticeably in the larger diameter of its conidia, for these bodies measure, on

an average, about 1.9 μ in greatest width, whereas the average for the corresponding dimension in *A. stenospora* is about 1.4 μ. From the conidia of *A. macrospora* those of the present fungus differ in their lack of any evident tendency either toward distal bifurcation or toward evacuation of contents from one of the ends. Besides, they generally taper upward less markedly, so that despite some measure of intergradation with respect to dimensions, they present a rather different appearance because of their more typically filamentous or rod-like shape. Their outward resemblance to walking sticks suggests for the fungus a specific epithet compounded in part of a word meaning "staff."

Acaulopage baculispora sp. nov.

Mycelium effusum; hyphis continuis, incoloratis, filiformibus, parce ramosis, plerumque 1–2 μ crassis, ad animalia minuta inhaerentibus, pelliculam eorum perforantibus, haustorium (interdum 2 haustoria) intus evolventibus quod protoplasma exhaurit; haustorio pedicellato, pedicello vulgo 2–5 μ longo, 0.6–1 μ crasso, apice abrupte latescente, bis usque quater repetite bifurco, ita 4–16 (interdum plures) ramulos assumentes divaricatos 2–12 μ longos, 1–1.4 μ crassos ferente. Sterigmata inter se saepe 5–50 μ distantia, erecta, plerumque 1–3.5 μ alta, basi 1–1.5 μ crassa, sursum attenuata, apice circa 0.6 μ vel 0.7 μ crassa, unicum conidium ferentia; conidiis erectis, incoloratis, aliquid filiformibus, 21–62 μ longis, 1.8–2.1 μ latis, utroque parvulum attenuatis, apice abrupte rotundatis. Hyphae zygosporiferae irregulariter flexuosae, ambae interdum ex duabus hyphis mycelii exeuntes sed saepissime altera ex hypha mycelii altera ex conidio germinanti oriunda, quisque gametangium vulgo 10–20 μ longum, 1.2–2.5 μ crassum ferens. Zygosporangia primo levia, sphaeroidea, plerumque 7–12 μ crassa, membrana eorum in maturitate circa zygosporam laxe collapsa; zygospora aliquantum flavida, globosa, 6–11 μ crassa, valde verrucosa, membrana ejus 1–2.2 μ crassa, cellulam viventem 3.5–6.5 μ crassam circumdante.

Amoebas 20–40 μ latas capiens consumensque habitat in herbis coacervatis putrescentibus prope Mayo, Maryland.

Mycelium spreading; vegetative hyphae aseptate, colorless, filamentous, sparingly branched, mostly 1–2 μ wide, adhering to minute animals, penetrating the pellicle of each animal thus captured, and intruding a haustorium (sometimes 2 haustoria) to appropriate the protoplasmic contents; haustoria pedicellate, the pedicel commonly 2–5 μ long and 0.6–1 μ wide, abruptly enlarging and bifurcating 2 to 4 times and thus bearing 4 to 16 (sometimes more) divergent assimilative branches 2–12 μ long and 1–1.4 μ wide. Sterigmata arising from procumbent hyphae often at intervals of 5–50 μ, usually 1–3.5 μ high, 1–1.5 μ wide at the base, tapering upward to a

width of 0.6–0.7 μ at the tip, whereon is borne erectly a single conidium. Conidia colorless, somewhat filamentous, 21–62 μ long, 1.8–2.1 μ wide, tapering only slightly toward both ends, abruptly rounded at the tip, when detached somewhat rounded at the base. Zygophoric hyphae often irregularly flexuous, both of a pair occasionally arising from separate mycelial filaments, but much more often only one of them arising from a mycelial filament, the other being supplied from a germinating conidium, each furnishing a gametangium commonly 10–20 μ long and 1.2–2.5 μ wide; zygosporangium formed usually 5–10 μ from place of union between gametangia, at first smoothly subspherical and mostly measuring 7–12 μ in diameter, but its envelope at maturity collapsing loosely about the zygospore; the latter somewhat yellowish, globose, 6–11 μ in diameter, boldly verrucose, having a wall 1–2.2 μ thick which surrounds a living cell 3.5–6.5 μ in diameter.

Capturing and consuming a species of *Amoeba* commonly 20–40 μ wide it occurs in heaped decaying herbaceous plants near Mayo, Maryland.

In examining young sexual reproductive apparatus of *Acaulopage baculispora* (FIG. 2, *I, e*) as well as of other small members of the Zoöpagaceae I have been unable to see in some conjugating elements any cross-wall so placed that it might be considered to delimit a gametangium. While failure to observe a delimiting septum may often be held attributable to optical difficulties frequent in the study of minute objects, the possibility is not to be dismissed that in some instances a special delimiting septum may be absent. The function of such septa in the Zoöpagaceae seems a little problematical since in most members they evidently do not isolate the protoplasm of the gametangium from that of the adjoining portion of mycelial branch or adjoining portion of conidium. In most members of the family, as has been intimated previously (**11**: 26), a pair of conjugating gametangia together contain only a rather small portion of the protoplasmic material eventually needed for the development of the zygosporangium and zygospore; so that of necessity the larger portion must enter the fusion cell after cross-walls have been formed. It may be presumed, therefore, that in the beginning, at least, the delimiting walls here very probably are not complete partitions, but like the centrally perforated cross-walls in the higher fungi (**1**: 75–167), have an aperture large enough

for slow movement of protoplasm—a feature which assuredly need not be shared by the cross-walls laid down in the progressive evacuation of mycelial hyphae and conidia (FIG. 2, *A*). If the apertures inferred from the sequence of developmental events should be more or less variable with respect to size, it appears possible, in view of the narrowness of hyphae in most Zoöpagaceae, that now and then a wall might for a time consist of a peripheral annular thickening so minute as to escape detection. Indeed, it is conceivable that with very narrow hyphae the measure of seclusion ordinarily provided by delimiting septa might perhaps be available here and there without any special partitions being required.

A SPECIES OF ACAULOPAGE PRODUCING ELLIPSOIDAL CONIDIA WITH LONG APICAL APPENDAGES

A maizemeal-agar plate culture which after being overgrown by *Pythium helicoides* Drechsl. had been further planted with decaying leaves collected on August 20, 1946, from the floor of deciduous woods near Mayo, Maryland, showed in eighteen days a species of *Acaulopage* reminiscent of the two forms I described earlier under the binomials *A. ceratospora* (**2**: 193–195) and *A. cercospora* (**5**: 371–374). Its scanty mycelium consisted of colorless, aseptate, sparingly branched hyphae measuring for the most part slightly less than 1 μ in width (FIG. 3, *A–D*). To these hyphae were found attached at irregular intervals small specimens of *Amoeba* (FIG. 3, *A, a–f; B, a–c; C, a*) varying in diameter from 6 to 21 μ when drawn into a somewhat rounded form. As the captured animals, like many other of the more minute *Amoebae* taken by predaceous hyphomycetes, seemed rather lacking in distinctiveness, they could not be satisfactorily identified. They were each enveloped by a very delicate pellicle, and they bore frequently a variable number of bacteria, which sometimes were visibly surrounded by small digestive vacuoles (FIG. 3, *A, d–f*). Within the larger specimens a globose or slightly elongated body measuring commonly 2.5 to 3 μ in total length and 2 to 2.5 μ in width (FIG. 3, *A: a, n; c, n; e, n*) seemed interpretable as a nucleus with a relatively large endosome. Many of the better developed captives (FIG. 3, *A, d, e*) were found invaded by a basally ramified bush-like haustorium composed of three or four assimilative branches

having about the same width as the mycelial hyphae. The smaller captives were often found invaded by only a single assimilative branch (FIG. 3, *C, a*). The delicate phycomycete was likewise restricted to intrusion of a single assimilative branch when, as occasionally happened, one of the larger specimens (FIG. 3, *A, f*), soon after being captured, was invaded adventitiously by a robust branch extended from a neighboring mycelial filament (FIG. 3, *A, g*) of the nematode-capturing hyphomycete *Dactylaria thaumasia* Drechsl. (**6**: 518–523) also present in the culture. Loss of protoplasm continued in all invaded animals until at a final stage of depletion the contents of the haustorium were withdrawn backward into the parent hypha; whereupon the empty tubular membranes soon faded from sight, together with the empty pellicle surrounding them.

Thus amply nourished on living animals the delicate phycomycete gave rise here and there to comparatively robust conidia. In initiating asexual reproduction the mycelial hyphae would put forth short branches that soon widened out rather abruptly to form an erect elongated-ellipsoidal part often about 15 μ long and from 5 to 6 μ wide. Through continued but markedly narrowed growth at its apex this swollen part came to be surmounted by a distal beak commonly 20 to 37 μ long and 1.3 to 1.9 μ wide (FIG. 3, *B, d; C, b*). At first the erect structure was filled uniformly with protoplasm throughout its length (FIG. 3, *B, d*). Later the beak began to show a vacuolated condition (FIG. 3, *C, b*); and this condition became more and more pronounced until all the protoplasmic material had been withdrawn from the beak into the swollen part below. A retaining wall was then formed at the base of the empty tubular membrane. At about the same time, if not earlier, another cross-wall was laid down between the swollen part and its supporting stalk (FIG. 3, *D, a; E*) which meanwhile, like the apical beak, had been emptied of contents. At this cross-wall separation took place on slight disturbance. Eventually numerous conidia consisting individually of an elongated-ellipsoidal living cell together with an empty tubular apical appendage were found strewn about in some abundance (FIG. 3, *F, a–z; G, a–e*). These detached conidia germinated rather freely by extending, often from a position close to the basal hilum (FIG. 3, *H*), a delicate vegetative

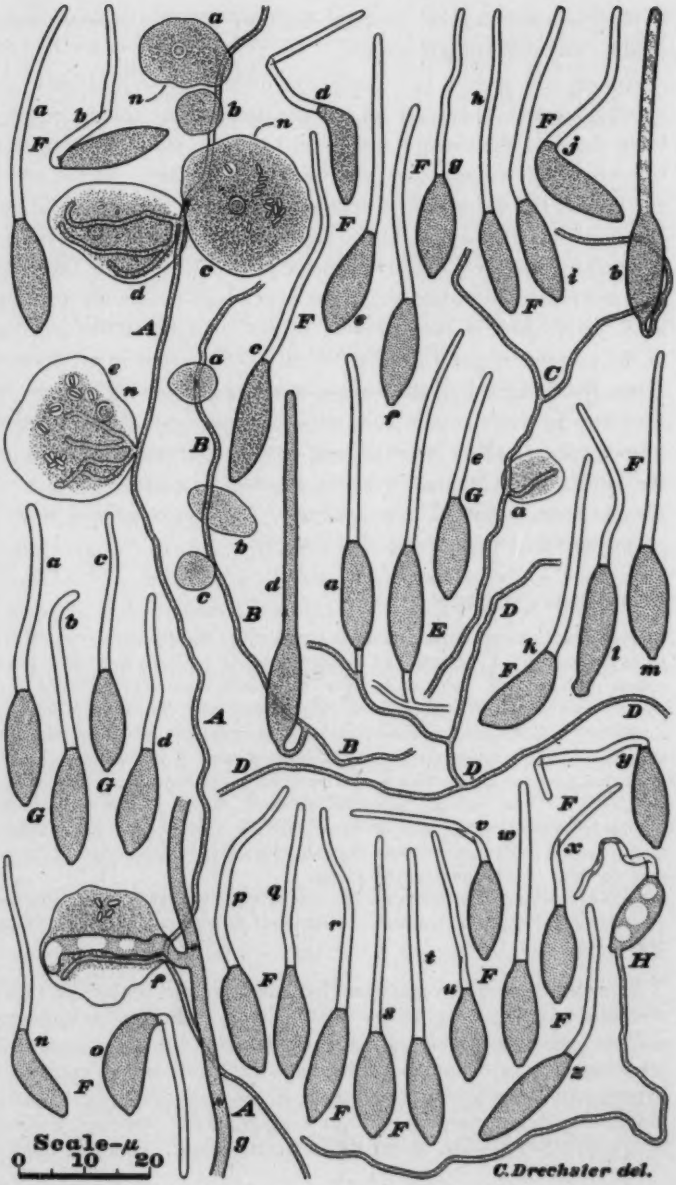

FIG. 3. *Acaulopage gyrinodes.*

hypha evidently capable of capturing any suitable *Amoeba* that might come into contact with it.

Among the species of *Acaulopage* hitherto described only *A. ceratospora* and *A. cercospora* are distinguished by conidia resembling those of the present fungus in having a swollen living cell surmounted by a single narrower tubular appendage. In the present fungus the living cell is scarcely two-thirds as long as in *A. ceratospora*. The apical appendage, too, is shorter than in *A. ceratospora* in approximately the same proportion, besides tapering less pronouncedly; while the basal appendage commonly present in *A. ceratospora* is absent here. By way of contrast the conidia of the present fungus exceed those of *A. cercospora* in all dimensions; the living cell in the former being longer and wider than in the latter by more than a half, while the apical appendage in the former appears about twice as long and three times as wide as in the latter. The fungus obviously represents a new species. A specific term meaning "like a tadpole" may prove helpful in recalling the distinctive shape of its conidia.

Acaulopage gyrinodes sp. nov.

Mycelium sparsum; hyphis continuis, incoloratis, filiformibus, parce ramosis, plerumque 0.8–1 μ crassis, ad animalia minuta inhaerentibus, pelliculam eorum perforantibus, haustorium intus evolventibus quod protoplasma exhaurit; haustorio interdum in ramo simplici constante, interdum arbusculiformi tum in 2–4 ramis consistente; ramis assumentibus 5–25 μ longis, 0.8–1 μ latis. Conidia incolorata, erecta, ex ramulis saepe 2–10 μ longis oriunda, in duabus partibus constantia; pars supera vacua, plerumque 20–37 μ longa, basi 1.3–1.9 μ lata, sursum parvulum attenuata, apice 1–1.6 μ lata et ibi abrupte rotundata, saepe plus minusve marcida vel collapsa; pars infera protoplasmatis repleta, plerumque elongato-ellipsoidea, vulgo recta sed interdum curvata, 14–20 μ longa, 4–6.5 μ lata.

Amoebas saepe 6–21 μ latas capiens consumensque habitat in foliis arborum (ex magna parte *Quercus, Corni, Liriodendri, Aceris*) putrescentibus prope Mayo, Maryland.

Mycelium sparse; vegetative hyphae aseptate, colorless, filamentous, sparingly branched, 0.8–1 μ wide, adhering to minute animals, penetrating the pellicle of each animal thus captured, and intruding a haustorium to appropriate the protoplasmic contents; haustorium sometimes consisting of a single assimilative branch, but more often consisting of 2 to 4 assimilative branches, 5–25 μ long and 0.8–1 μ wide, in bush-like arrangement. Conidia color-

less, erect, arising singly from branches 2–10 μ long, each spore consisting of two parts: an upper empty tubular part, 20–37 μ long, 1.3–1.9 μ wide below, 1–1.6 μ wide above, bluntly rounded at its tip, often becoming more or less collapsed; and a lower living part usually of elongated-ellipsoidal shape, usually straight but occasionally somewhat curved, 14–20 μ long, and 4–6.5 μ wide.

Capturing *Amoebae* often 6–21 μ wide it occurs in decaying leaves of deciduous trees (including species of *Quercus, Cornus, Liriodendron,* and *Acer*) near Mayo, Maryland.

A STYLOPAGE PRODUCING DISTALLY APPENDAGED CONIDIA ON TALL SLENDER CONIDIOPHORES

Among the members of the genus *Stylopage* that have been set forth as subsisting by capture of *Amoebae,* the two species I described under the binomials *S. araea* (**2**: 199–201) and *S. rhynchospora* (**8**: 394–397) are distinctive especially by reason of the unusual height—commonly about 200 μ—of the slender conidiophores on which they produce singly their much wider ellipsoidal conidia. In the stature of their conidiophores as well as in the shape of their conidia these species have supplied a transition to the nematode-capturing forms *S. hadra* Drechsl. (**3**) and *S. leiohypha* Drechsl. (**4**) whose robust dimensions might otherwise have made them seem rather alien to the predaceous Zoöpagaceae. A third species manifestly belonging in the same category with *S. araea* and *S. rhynchospora* was recently obtained from leaf mold kindly collected by A. W. Rakosy in oak woods near Frederick, Maryland, on January 15, 1947. The new fungus came to light in several maizemeal-agar plate cultures which after being overgrown by *Pythium irregulare* Buisman had been further planted with small quantities of the forest detritus. When first observed twenty-two days after the leaf mold was added, it was already sporulating somewhat extensively on the agar substratum and must therefore have grown out from the partly decayed oak-leaf fragments nearby, several days—probably between five and ten days—earlier.

The scanty mycelium of the new fungus consisted of meagerly branched aseptate hyphae, mostly about 1 μ wide, along which

specimens of *Amoeba* commonly measuring 10 to 20 μ across were found attached at variable intervals (FIG. 4, *A–D*). Most of the captured animals were wholly or in large part depleted of their protoplasm; this depletion having evidently been accomplished by rangy bush-like haustoria consisting individually of two to five assimilative branches of about the same width as the parent hyphae. Mycelial filaments on the surface of the agar substratum gave rise in scattered positions to erect solitary conidiophores (FIG. 4, *E, a*) which from a basal diameter slightly in excess of 1 μ tapered upward very gradually until at a height of approximately 150 μ, where their width often measured only 0.6 or 0.7 μ, they expanded into a rather massive elongated-ellipsoidal or somewhat fusiform part with a narrowing apical beak (FIG. 4, *E, b*). Later a cross-wall was formed at the base of the swollen part, and the beak was evacuated of contents; so that the individual conidiophore (FIG. 4, *F, a*), now delimited distally, came to support aloft a fusiform or elongated-ellipsoidal conidium (FIG. 4, *F, b*) with a membranous apical appendage. Frequently here, as also in *Stylopage araea*, a short distal portion of the conidiophore was seen to be empty of protoplasmic contents while the conidium was still attached (FIG. 4, *F, a*). In some instances where disarticulation had probably been hastened by the disturbance entailed in mounting material under a coverglass, denuded conidiophores were found filled with protoplasm throughout their length (FIG. 4, *G*); and prematurely detached conidia likewise were seen filled to the tip of the beak (FIG. 4, *H, I*). Where the conidia had matured normally, however, the beak seemed regularly represented by an empty tubular appendage often from one-fourth to one-third as long as the living cell. The appendages were clearly visible, though usually in a badly collapsed condition, when undisturbed cultures were examined microscopically by means of a dry objective. In moist preparations they occasionally were revealed with distinctness and in their original shape (FIG. 4, *J–N*), but more often in such mounts their diaphanous character and badly collapsed condition made them difficult to see even though the truncate distal end of the living cell left no doubt as to their presence (FIG. 4, *O, a–z; P, a–z; Q, a–h*). Germination of detached conidia was found to take place by emission of a germ hypha close to the basal hilum (FIG. 4,

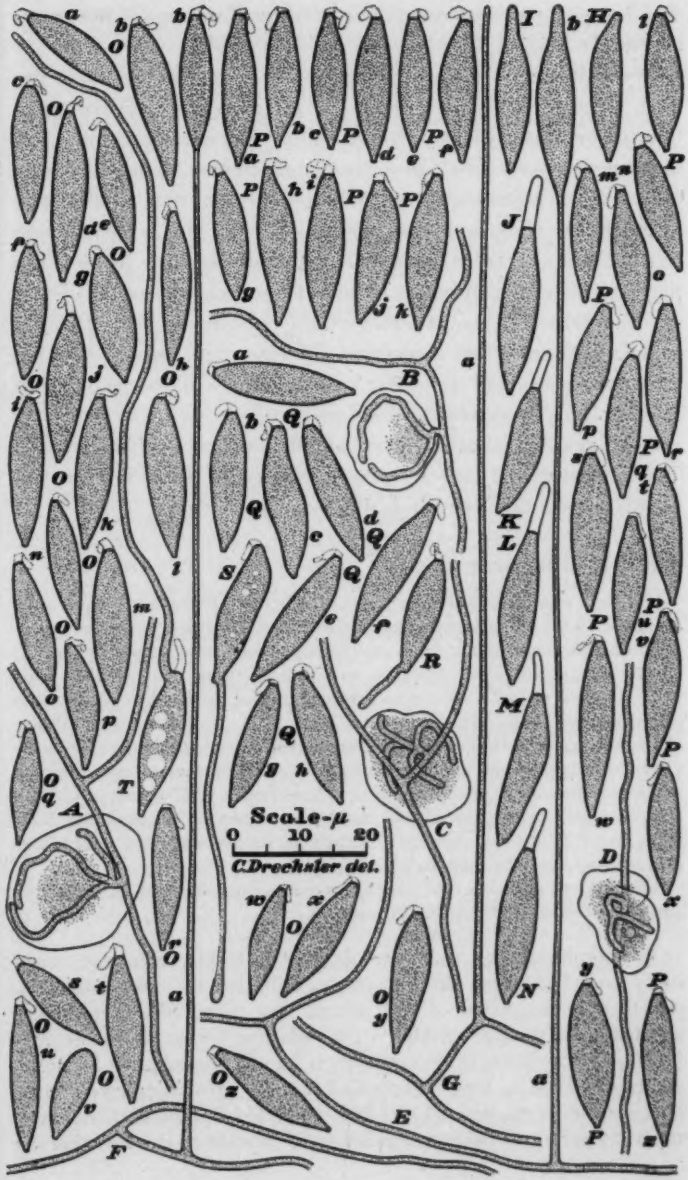

FIG. 4. *Stylopage rhionacra.*

R, S) or, in other instances, close to the wall setting off the empty appendage (FIG. 4, *T*).

Development of secondary conidia on germ conidiophores put forth erectly from fallen conidia—a type of repetitional reproduction frequent in *Stylopage rhynchospora*—has never been observed in the new fungus. Nor have conidia of the new fungus ever been observed bearing at the tip any adhesive material such as is present in the yellow globular masses whereby conidia of *S. rhynchospora* often cohere apically in pairs. Owing mainly to their lesser width they are of noticeably more slender shape than the conidia of *S. rhynchospora*. In comparison with the consistently unappendaged conidia of *S. araea* they show little difference with respect to width, but because of their generally greater length offer, again, conspicuously more slender proportions.

A term compounded of two words meaning "withered" and "tip," respectively, may serve conveniently as an epithet for the fungus in recalling its collapsed conidial appendage.

Stylopage rhicnacra sp. nov.

Mycelium sparsum; hyphis continuis, incoloratis, filiformibus, parce ramosis, saepe 0.9–1.3 μ crassis, ad animalia minuta inhaerentibus, pelliculam eorum perforantibus, haustorium intus evolventibus quod protoplasma exhaurit; haustorio arbusculiformi, in aliquot (saepe 2–5) ramis assumentibus vulgo 10–20 μ longis, circa 1 μ crassis constante. Hyphae fertiles incoloratae, simplices, erectae, saepe 140–175 μ longae, basi 1–1.3 μ crassae, sursum leviter attenuatae, apice 0.6–0.7 μ crassae, ibi unicum conidium ferentes; conidiis vulgo in cellula viventi et appendice vacua terminali consistentibus: cellula viventi incolorata, elongato-ellipsoidea vel fusiformi, 17–27 μ longa, 4.5–6.5 μ lata, basi acutula, sursum aliquid applanata; appendice vacua vulgo 5–8 μ longa, basi 1.2–1.8 μ lata, nunc cylindracea nunc sursum attenuata, apice rotundata, saepissime valde marcida et collapsa.

Amoebas saepe 10–20 μ latas capiens consumensque habitat in foliis *Quercus* putrescentibus prope Frederick, Maryland.

Mycelium scanty; vegetative hyphae aseptate, colorless, sparingly branched, often 0.9–1.3 μ wide, adhering to minute animals, penetrating the pellicle of each animal thus captured and intruding a haustorium to appropriate its protoplasmic contents; haustorium bush-like, consisting of several (often 2 to 5) assimilative branches commonly 10–20 μ long and about 1 μ wide. Conidiophores colorless, simple, erect, often 140–175 μ long, 1–1.3 μ wide at the base, tapering very gradually upward, 0.6–0.7 μ wide at the tip whereon

is borne a single conidium. Conidia consisting of a living cell and an empty terminal appendage—the living cell colorless, elongate-ellipsoidal or somewhat spindle-shaped, 17–27 μ long and 4.5–6.5 μ wide, somewhat acute at the narrow basal end, roundly truncate at the wider distal end—the empty terminal appendage commonly 5–8 μ long and 1.2–1.8 μ wide at the base, sometimes virtually cylindrical but at other times tapering upward perceptibly, always rounded at the tip, very often withered and collapsed.

Capturing and consuming *Amoebae* 10 to 20 μ wide it occurs in decaying leaves of *Quercus* near Frederick, Maryland.

DIVISION OF FRUIT AND VEGETABLE CROPS AND DISEASES,
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EXPLANATION OF FIGURES

FIG. 1. *Acaulopage baculispora*, drawn to a uniform magnification with the aid of a camera lucida; × 1,000 throughout. *A, B*, Portions of mycelial hyphae from each of which a young haustorium has been intruded into a captured specimen of the *Amoeba* sp. most commonly taken as prey. *C–E*,

Branched portions of mycelium from each of which a fairly well developed haustorium has been intruded into a captured specimen of the *Amoeba* sp. most frequently serving as prey. *F, G,* Branched portions of mycelium from each of which two haustoria have been intruded into a separate specimen of the *Amoeba* sp. most frequently taken as prey; in each instance one haustorium, *a,* is in young condition whereas the other, *b,* is in further developed condition. *H, I,* Portions of mycelial hyphae, each with a richly branched haustorium in a captured *Amoeba* that has been very largely expropriated of its protoplasm. *J, K,* Portions of hyphae possibly belonging to the same predaceous fungus; from each portion a young haustorium has been intruded into a specimen of an *Amoeba* sp. that was not frequently observed taken as prey. *L,* Young unit of sexual reproductive apparatus produced by two mycelial hyphae, *a* and *b;* these hyphae have given off branches bearing gametangia whose conjugation is being followed by development of a zygosporangium, *c. M,* Unit of sexual reproductive apparatus contributed jointly by a germinating conidium, *a,* and a mycelial hypha, *b;* the paired gametangia have fused, and a zygosporangium, *c,* is beginning to develop in the gametangium supplied from the mycelial hypha. *N,* Two units of sexual reproductive apparatus formed through interaction of a germinating conidium, *a,* with two mycelial hyphae, *b* and *c;* the conidium has given rise to two gametangia on separate germ hyphae; these gametangia have conjugated with two others borne separately on branches given off by the mycelial hyphae; in the latter gametangia the zygosporangia *d* and *e* are being formed. *O,* Mature unit of sexual reproductive apparatus contributed jointly from a conidium, *a,* and a mycelial hypha, *b;* the zygosporangium *c,* which contains a mature zygospore, was formed in the gametangium supplied from the mycelial hypha. *P,* Mature zygospores, *a–p,* each loosely surrounded by the slightly collapsed zygosporangial envelope; the attachment of the envelope to the empty tubular membranes of the gametangia is shown in most instances. (*n,* nucleus of captured *Amoeba; s,* cross-wall delimiting gametangium proximally; *u,* place of union between paired gametangia; *v,* contractile vacuole of captured *Amoeba.*)

FIG. 2. *Acaulopage baculispora,* drawn to a uniform magnification with the aid of a camera lucida; × 1,000 throughout. *A,* Portion of branched prostrate hypha active in asexual reproduction: *a,* erect conidium in early stage of growth; *b,* erect conidium probably full grown or nearly full grown, though not yet delimited at its base; *c–g,* five sterigmata, each bearing erectly a basally delimited conidium. *B,* Portion of branched prostrate mycelial hypha bearing five sterigmata, *a–e,* each bearing an erect conidium. *C,* Portion of prostrate mycelial hypha bearing five denuded sterigmata, *a–e. D* (*a–z*), *E* (*a–w*), Detached conidia, showing usual variations in size and shape. *F,* Detached conidium germinating obliquely from its distal end. *G,* Young unit of sexual reproductive apparatus contributed from a germinating conidium, *a,* and a mycelial hypha, *b. H,* Somewhat older unit of sexual reproductive apparatus likewise contributed from a germinating conidium, *a,* and a mycelial hypha, *b; c,* young growing zygosporangium. *I,* Two units of sexual reproductive apparatus resulting from conjugation of two gametangia supplied from the germinating conidium *a* with two other gametangia supplied from the mycelial hypha *b;* in the latter two gametangia the zygo-

sporangia *c* and *d*, respectively, are being formed; one of these gametangia, *e*, is not yet visibly delimited by a cross-wall. *J*, Mature zygospores, *a–l*, each loosely surrounded by the zygosporangial envelope (*s*, cross-wall delimiting gametangium proximally; *u*, place of union between paired gametangia).

FIG. 3. *Acaulopage gyrinodes*, drawn to a uniform magnification with the aid of a camera lucida; × 1,000 throughout. *A*, Portion of mycelial hypha with six captured *Amoebae*, of which three, *a–c*, have not yet been invaded, whereas the others, *d–f*, have been partly depleted of their protoplasm by haustorial branches intruded into them in numbers of three, four, and one, respectively; one captive, *f*, has been further invaded by a stout branch from a mycelial hypha, *g*, of *Dactylaria thaumasia*; *n*, nucleus of captured animal. *B*, Prostrate mycelial hypha on which three small *Amoebae*, *a–c*, are held captive; the hypha has given rise on a short lateral branch to an erect conidium, *d*, which though full grown has not yet been delimited at its basal end, and is still uniformly filled with protoplasm throughout its apical prolongation. *C*, Portion of prostrate mycelial hypha which has extended an assimilative branch into a small captured *Amoeba*, *a*, and has produced an erect conidium, *b*, from whose vacuolated apical prolongation the protoplasmic contents are being withdrawn. *D*, Portion of prostrate mycelium bearing a mature erect conidium, *a*, whose ellipsoidal living cell is delimited basally from the empty stalk and distally from the empty elongate apical appendage. *E*, Empty portion of prostrate hypha bearing a mature appendaged conidium on a short empty stalk. *F* (*a–z*), *G* (*a–e*), Mature detached conidia showing variations in size and shape of the living cell as well as of the empty apical appendage. *H*, Detached conidium that has put forth a germ hypha obliquely from a position near its base.

FIG. 4. *Stylopage rhicnacra*, drawn to a uniform magnification with the aid of a camera lucida; × 1,000 throughout. *A–D*, Portions of mycelial hyphae from each of which a branched haustorium has been extended into a captured *Amoeba*; all the captives have been largely expropriated of their protoplasmic contents. *E*, Portion of prostrate hypha from which has been sent up a slender erect conidiophore, *a*, that is still continuous distally with the conidium, *b*, whose apical prolongation is still filled uniformly with protoplasm. *F*, Portion of prostrate mycelium from which has been sent up an erect conidiophore, *a*, bearing a mature conidium, *b*, that is provided with an empty apical appendage. *G*, Portion of prostrate hypha with an erect conidiophore, *a*, not supporting a conidium. *H*, *I*, Detached conidia in which the apical beak is still filled with protoplasm. *J–N*, Detached conidia wherein the empty apical appendage has not collapsed or shriveled. *O* (*a–z*), *P* (*a–z*), *Q* (*a–h*), Random assortment of detached conidia showing usual variations in size and shape of the living cell; the apical appendage here being in varying degree collapsed or shriveled, and often barely discernible. *R*, *S*, Detached conidia, each with a germ tube arising in close proximity to its base. *T*, Detached conidium with a germ hypha arising in close proximity to its distal end.

A COMMON ANTIGENIC FACTOR IN DIFFERENT SPECIES OF SPOROTRICHUM

H. I. Lurie [1]

Dodge (**8**) describes six human and one equine species of *Sporotrichum*. The human ones are *S. Grigsbyi, S. Fonsecai, S. asteroides, S. cracoviense, S. Carougeaui, S. Jeanselmei* and *S. Schenckii* and the equine species is *S. equi*. He recognizes four varieties of *S. Schenckii*, namely vars. *Beurmanni, Greconis, Councilmani* and *Fioccoi*. His key to the species is based on liquefaction of gelatine, coagulation of milk, and pigment formation. However, he states that "the delimitation of species in this genus is very difficult. Color, which has often been used not only to separate species but even genera, has been shown by Davis (**6**) to be variable. Practically all strains that he tried formed pigment when first isolated. . . . Albino strains frequently occur and remain fixed even after passage through an experimental animal. Meyer and Aird (**10**), after a study of eighteen strains, found some differences in carbohydrate fermentation, but these were not sufficiently constant or correlated with other characters to warrant separation into species." Davis (**5**) found that the production of chlamydospores was of no value in the differentiation of species.

Meyer (**9**) states that extensive bacteriologcial and serological experiments have proved the identity of the causative organisms in human and animal sporotrichosis, and that the pathogenicity of the equine strain for human beings was observed in an accidental laboratory infection. Davis (**4**) compared three human and two equine strains by their agglutination reactions and found no differences. Two of his human strains were isolated in America and

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classified as *S. Schenckii,* the third was received from Gougerot in Paris and classified as *S. Beurmanni.* The equine strains were supplied by Dr. Meyer of Philadelphia. In 1921 Davis (7), after a careful study of the original descriptions of *S. Schenckii* and *S. Beurmanni* and comparison of the cultures, concluded that they were identical and that the name *S. Schenckii* should have priority. He also states that *S. Jeanselmei* is probably identical with *S. Beumanni* but concedes that *S. Dori* and *S. Councilmani* are different. The former has been lost and the latter was unfortunately not available for this study.

The object of this investigation is to establish the antigenic identity of different strains of *Sporotrichum* after the method of Davis (4). The method he employed however was rather laborious. It required eight months to obtain a satisfactory titer in his rabbits. Recently Campbell (3) described a technique for culturing *Sporotrichum* in a "yeast" phase by growing the organism on cystine-agar at 37° C. This appeared to be an ideal method to obtain a concentrated suspension of spores free from mycelium for use as an antigen. Preliminary experiments proved this in fact to be the case. The method finally adopted is described below.

Sixteen strains of *Sporotrichum* were selected. Their origins and classifications are as follows:

No. 1. *S. Schenckii,* human case, Duke Hospital, N. Carolina.
No. 2. *S. Schenckii,* human case, Duke Hospital, N. Carolina.
No. 3. *S. Schenckii,* human case, Temple, Texas.
No. 4. *S. Schenckii,* human case, isolated from spleen, Cincinnati, Ohio.
No. 5. *S. Schenckii,* human case, New York.
No. 6. *S. Schenckii,* human case, Virginia.
No. 7. *S. Schenckii,* human case, Indiana.
No. 8. *S. Schenckii,* human case, Mexico.
No. 9. *S. asteroides,* Splendore, Westerdijk, Baarn.
No. 10. *S. Beurmanni,* M. and Ram, Westerdijk, Baarn.
No. 11. *Rhinocladium Beurmanni,* Langeron, Paris.
No. 12. *Rhinocladium equinum,* Sabouraud's collection, Langeron, Paris.
No. 13. *S. Beurmanni,* isolated from mine timber, South Africa.
No. 14. *S. Beurmanni,* isolated from mine timber, South Africa.
No. 15. *S. Beurmanni,* human case contracted in a mine, South Africa.
No. 16. *S. Beurmanni,* human case contracted in a mine, South Africa.

Numbers 1 to 12 were supplied by Dr. Conant and numbers 13 to 16 were supplied by Brown, Weintroub and Simpson (2) of the

Transvaal Chamber of Mines Timber Research Laboratory. The color of the cultures varied from creamy-white to dark brown, almost black.

All cultures were first grown on Sabouraud's dextrose-agar at room temperature for two weeks. They were then transferred to Difco Cystine-agar slopes and incubated at 37° C. for five to seven days, after which they were transferred to fresh Cystine-agar slopes and incubated for a further five to seven days. At the end of this period the majority of cultures appeared to be quite "yeasty." However, a few required further transfers and one (no. 12) required to be passed through a mouse before a "yeasty" growth was obtained. The growths from four slopes were transferred with a platinum loop to approximately 30 cc. of sterile isotonic saline. This was sufficient to give a milky suspension of spores. Where it was necessary to remove pieces of agar or large clumps of spores the suspension was filtered through several layers of gauze. The spore suspensions were then killed by heating 60° C. for two hours. The density of each spore suspension was measured by centrifugation in Hopkin's tubes and each adjusted by the addition of saline to a concentration of 1 : 200. From this a suspension of 1 : 1,000 was made. The former was used for the injection of rabbits, the latter for the subsequent agglutination tests. All suspensions were tested for sterility in the usual manner before use.

Four strains were selected for the production of antiserum, namely Nos. 1, 3, 9 and 12, two being classified as *S. Schenckii*, one as *S. asteroides* and one as *Rhinocladium equinum*.

The rabbits were injected intravenously according to the schedule recommended by Benham (1) for Monilias. On the first day 1 cc. was given, 2 cc. on the second day and 5 cc. on the third day. The same doses were repeated a week later, and the rabbits bled one week after the last injection. One rabbit required a further three injections before a satisfactory titer was obtained. The final titers were between 1 : 640 and 1 : 1,280.

Agglutination reactions were carried out with all sixteen strains using each of the four antisera. An amount of 0.5 cc. of a 1 : 1,000 suspension of the antigen was added to 0.5 cc. of antiserum in dilutions ranging from 1 : 10 to 1 : 2,560, the mixtures then shaken and

the racks placed in a waterbath at 37° C. overnight. A control of 0.5 cc. antigen suspension with 0.5 cc. saline was used in each case to detect auto-agglutination and the results were read macroscopically. Where the last tube showing agglutination displayed complete clumping the result was designated by a +; where the last tube showed only partial agglutination the result was designated by a ±. A summary of the results is given in Table 1.

TABLE 1

Strain	Titer of Agglutination with Antiserum			
	1	3	9	12
1	+1280	+ 320	+ 640	+ 640
2	±1280	+ 640	± 640	+1280
3	±1280	±1280	±1280	+ 640
4	± 640	+ 320	± 640	+1280
5	± 640	±1280	± 640	± 640
6	+1280	+1280	+1280	+ 640
7	± 640	+ 320	± 640	± 640
8	± 640	+ 320	± 640	+ 640
9	± 320	± 640	+ 640	±1280
10	+ 640	± 640	± 640	±1280
11	+ 640	± 640	+ 640	+ 640
12	+1280	± 640	±1280	+1280
13	± 640	+ 640	±1280	± 640
14	±1280	+ 640	±1280	+1280
15	±1280	±1280	±1280	+1280
16	± 640	+ 320	+ 320	±1280

Each antiserum agglutinated all sixteen strains to approximately equal titers. In no case was any one strain agglutinated in lower titer than the other strains by all the antisera. All sixteen strains examined therefore have a common antigenic factor.

In order to confirm these findings absorption experiments were carried out. Antiserum no. 3 was absorbed with strain no. 9 for a short time (1 cc. antiserum with 4 cc. 1 : 200 antigen for 2 hours at 37° C.). The antiserum-antigen mixture was then centrifuged and the supernatant fluid used for agglutinating all sixteen strains. The results are shown in Table 2. The titer of the antiserum was slightly lowered for all sixteen strains and to an approximately equal degree. Antiserum no. 1 was absorbed with strain no. 9 for a longer period (18 hours) in the same proportions as above. The supernatant fluid was again used for agglutinating all sixteen

TABLE 2

Strain	Original Titer with Antiserum no. 3	Titer after Absorption with Strain no. 9 (2 hours)
1	+ 320	±160
2	+ 640	±320
3	±1280	±320
4	+ 320	+160
5	±1280	+320
6	+1280	±640
7	+ 320	±160
8	+ 320	+160
9	± 640	+160
10	± 640	+160
11	± 640	+320
12	± 640	±160
13	+ 640	±320
14	+ 640	+320
15	±1280	±320
16	+ 320	±160

strains. The results are shown in Table 3. There is an appreciable and equivalent drop in titer in each case. Similarly antiserum no. 12 was absorbed with strain no. 1. The results are shown in Table 4.

TABLE 3

Strain	Original Titer with Antiserum no. 1	Titer after Absorption with Strain no. 9 (18 hours)
1	+1280	+ 80
2	±1280	±160
3	±1280	±160
4	± 640	± 80
5	± 640	+ 80
6	+1280	+160
7	± 640	± 80
8	± 640	+ 80
9	± 320	±160
10	+ 640	± 80
11	+ 640	+ 80
12	+1280	+160
13	± 640	+ 80
14	±1280	±160
15	±1280	±160
16	± 640	± 80

Finally antiserum no. 12 was absorbed with varying proportions of a 1:2 suspension of strain no. 3. The smallest amount of antigen which had completely absorbed its own antibodies was determined. This was found to be equal proportions of antiserum and a 1:2 suspension of antigen. A further sample of no. 12 antiserum was then absorbed with this amount of strain no. 3.

TABLE 4

Strain	Original Titer with Antiserum no. 12	Titer after Absorption with Strain no. 1 (18 hours)
1	+ 640	±320
2	+1280	±320
3	+ 640	±320
4	+1280	+160
5	± 640	+160
6	+ 640	±320
7	± 640	±320
8	+ 640	±160
9	±1280	±320
10	±1280	+320
11	+ 640	±160
12	+1280	±320
13	± 640	±320
14	+1280	±320
15	+1280	±640
16	±1280	±320

The absorbed serum was then put up against all sixteen strains. The results are shown in Table 5. It will be seen that there has been practically complete absorption of the agglutinins for all sixteen strains.

TABLE 5

Strain	Original Titer with Antiserum no. 12	Titer after Complete Absorption with Strain no. 3
1	+ 640	—
2	+1280	—
3	+ 640	—
4	+1280	—
5	± 640	—
6	+ 640	—
7	± 640	—
8	+ 640	—
9	±1280	±20
10	±1280	—
11	+ 640	—
12	+1280	—
13	± 640	—
14	+1280	—
15	+1280	±20
16	±1280	±20

All sixteen strains (three species and one variety), therefore, contain a common antigenic factor.

SUMMARY

1. A simple technique for the preparation of *Sporotrichum* antigen, the immunization of rabbits and an agglutination test are described.

2. Sixteen different strains of *Sporotrichum* belonging to at least three species, originating in three continents and varying in color from creamy-white to dark brown, are compared by their serological reactions.

3. *S. Schenckii, S. Beurmanni, S. asteroides* and *Rhinocladium equi* originating in North and South America, Europe and South Africa all have a common antigenic factor.

4. This common antigenic factor is present irrespective of the degree of pigmentation of the fungus.

5. The *Sporotrichum* growing saprophytically on the timber in the mines in South Africa contains the same antigenic factor as the *Sporotrichum* isolated from the lesions of workers in these mines.

Further absorption experiments are being carried out in order to determine whether the antigenic structures of these species of *Sporotrichum* are identical or merely contain one common factor.

ACKNOWLEDGMENTS

I am indebted to the Duke University for their hospitality and for granting me the facilities to carry out this work; to Dr. Norman F. Conant for his invaluable advice and constant interest; to Miss Jean Y. Biser and Mr. William C. Council for the preparation of the media.

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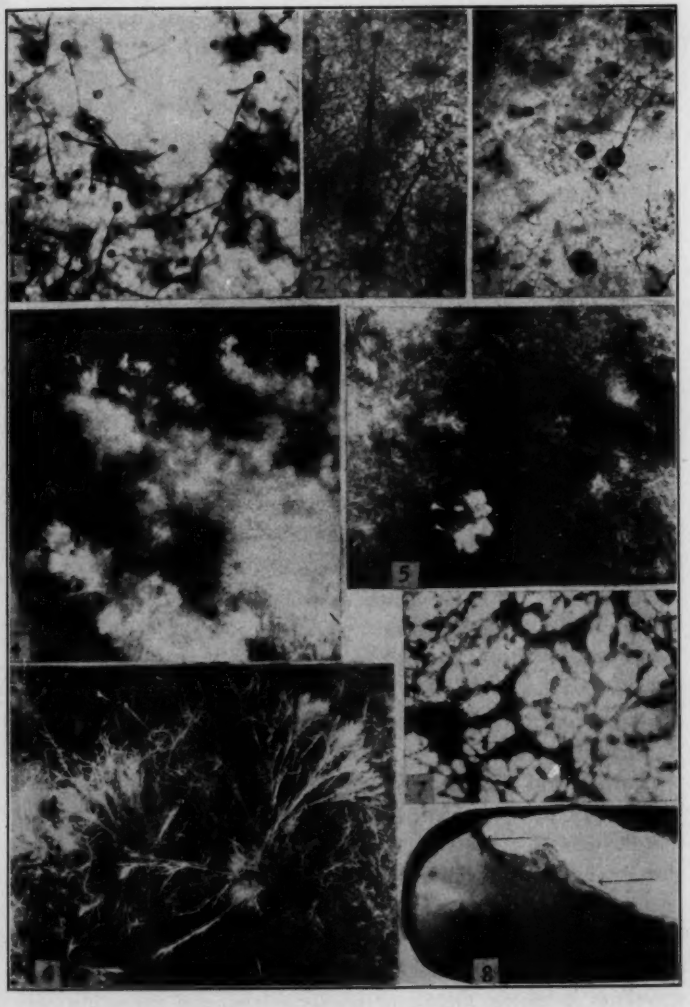
SPHAERONAEMELLA FIMICOLA (MARCHAL) EMEND.: SOME CHARACTERISTICS IN CULTURE

DOROTHY PEASE

(WITH 19 FIGURES)

It has been shown for several of the species which were originally assigned to the genus *Sphaeronaema* that the supposed pycnidia are in reality ascocarps (Schwarz, **1922**; Elliott, **1923**; Sartoris, **1927**; Dade, **1928**). These species were thereafter removed to the genus *Ceratostomella,* and later to *Ophiostoma* (*Ceratostomella*) (Sydow and Sydow), (Melin and Nannfeldt, **1934**). All of the species thus transferred produce black ascocarps. When Nannfeldt and Melin (**1934**) revised the genus *Ceratostomella* to *Ophiostoma* they suggested the probability that other speces of *Sphaeronaema* might eventually be found to belong in it; but these could be only such species as have black fruits. More recently Seeler (**1943**) found that the fruiting bodies of *Sphaeronaema Helvellae* were in reality ascigerous instead of pycnidial. This species produces light colored, fleshy ascocarps which exclude it from *Ophiostoma* (*Ceratostomella*); furthermore it has hyaline ascospores which exclude it from the dark-spored *Melanospora.* First described by Karsten as *Sphaeria Helvellae,* it was later made by him the type species for a new genus, when it became *Sphaeronaemella Helvellae* (Karsten **1884**). Jaczewski (**1898**) in his monograph on *Sphaeronaema* transferred the *Sphaeronaemellae* to *Sphaeronaema.* However, with the establishment of the ascigerous character of the fruits of *S. Helvellae* it became necessary to remove it from this genus, and Seeler (**1943**) has returned it to *Sphaeronaemella,* reëstablishing the earlier genus with an emended description which places it among the Ascomycetes in the Nectriaceae, near *Melanospora.* Thus a place has been made for other light fruited, hyaline spored species of *Sphaeronaema* which may prove in reality to be Ascomycetes.



FIGS. 1–8. *Sphaeronaemella fimicola.*

The present paper deals with such a light colored *Sphaeronaema* with hyaline spores, whose fruits are ascocarps, not pycnidia. It must therefore join Seeler's *Sphaeronaemella Helvellae* (Karsten). Our fungus is a saprophyte, found on squash, citron, and gourd which has begun to rot on the ground. Though visible growth may be suppressed by dry air or by other molds in the same environment, two asexual forms are sometimes found in the field. At the margin of the rotting area on the fruit delicate tufts of short unbranched hyphae, tipped with curving chains of conidia, erupt through the fruit rind, like a miniature setting of shrubs (FIG. 9). Toward the center of the rotted area the mold forms a weft of snow white mycelium traversed by heavy ropes of conidia which often collect in large moist clumps (FIG. 7). Sexual fruits have not been found in any of our field collections, but develop abundantly in moist chamber preparations and in cultures kept at about 15° C. (FIGS. 1, 2, 3).

CULTURES. This species grows well on agar which contains plant material if the humidity in the culture dish, and the water content of the agar, are both relatively high. Irrigating wells (Pease **1935**) have been used to provide these conditions.

MYCELIUM. Mycelial growth is white, the form of the growth varying with the environment. The hyphae are composed of uninucleate cells, 12–16 μ long, and average 2.4 to 3.2 μ in diameter. On sterile squash rind which has been kept thoroughly moist there are produced the delicate tufts of conidiophores which are sometimes seen in the field (FIG. 9). The aerial growth of mycelium with ropes and masses of conidia develops in high humidity and cool temperature, especially from mass conidial and mass ascospore inocula (FIG. 7). Heavy inoculaton of agar whch contains finely sieved squash rind and slightly lowered water content develops profusely branched white tufts of mycelium often producing no conidia (FIGS. 4, 5, 6). These are especially noticeable in single ascospore strains. Occasionally cultures develop in which the mycelium is sparse, mostly or wholly sterile, and spreading over the entire plate with no distinguishing form.

CONIDIA. These cells are oval, non-septate, with a single nucleus, commonly 12 × 5–6 μ (FIGS. 16, 17, 18). A longer somewhat curved septate conidium may very rarely be found. All conidia

germinate readily in water, producing from one to as many as six germ tubes. Conidial production varies with the habitat in which the inoculum is placed. Like species of *Ophiostoma* (*Ceratostomella*) described by Münch (**1907**), Clinton & McCormack (**1936**), Lagerberg, Lundberg, and Melin (**1927**), and Goidanich (**1935**), our species produces cephalosporial, verticillate, "stag-horn" (FIG. 14), and other types of growth. Conidia develop both terminally and laterally. The two most characteristic forms are probably those found in the field, chalaral (FIG. 9) and the long horizontal chains and ropes of conidia described by Münch (**1907**) for his blue-stain Ceratostomellae.

Two types of conidiophores described for various species of *Ophiostoma* and related fungi have not appeared in the hundreds of cultures made from conidial and ascospore inocula during a five year period of study. These are the graphial and penicilloid fructifications. (*Cf.* Münch **1907**, Schwarz **1922**, Grossman **1932**, and others.)

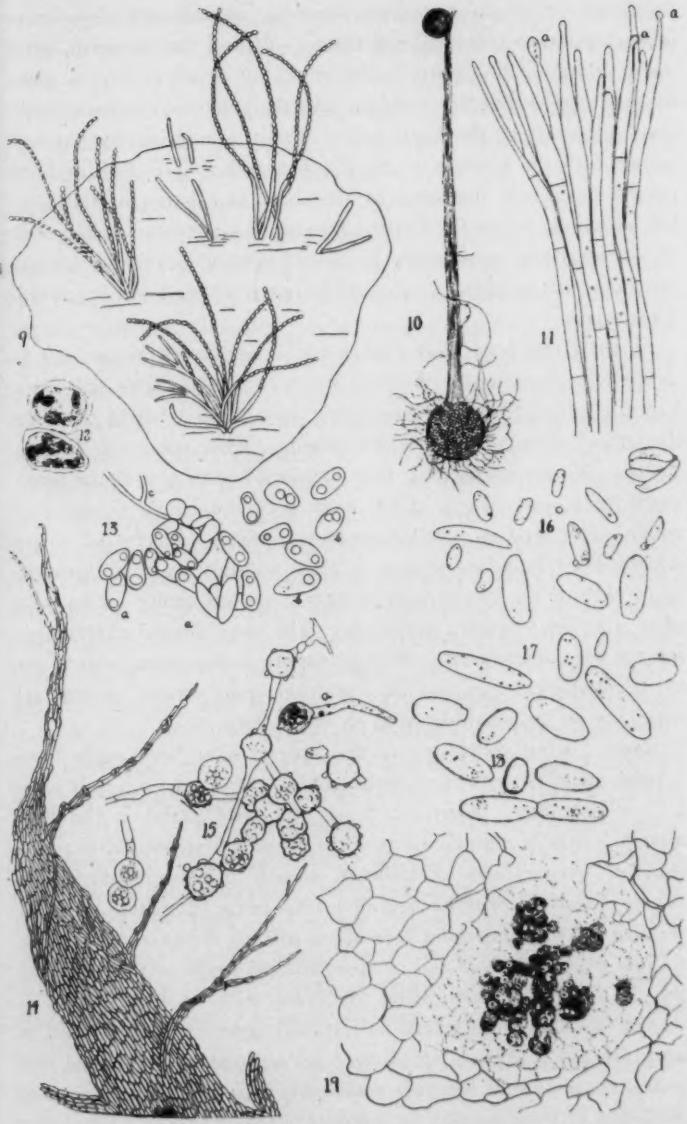
CHLAMYDOSPORES. These structures are conspicuous under the microscope in mycelia produced from mass conidial or mass ascospore inocula. High humidty and moisture and lower temperature favor them. Either terminal or intercalary cells enlarge to a diameter of 8 μ (extremes: 5.6 to 16 μ), develop a very thick hyaline mucilaginous wall which may remain smooth or bear large warty protuberances (FIG. 15). Their contents are large closely packed globules which are probably neither volutin nor oil, but react more like glycogen. Chlamydospores are especially abundant in the vicinity of the base of the ascocarp; they are often found inside the base of old recumbent beaks. Frequently they are accompanied by a slightly flattened adjacent cell, in appearance resembling a receptacle. They germinate readily in water mounts, producing one or more germ tubes.

YEAST-LIKE CELLS. In old cultures, kept moist and cool, yeast-like cells develop abundantly in the mycelial mat in direct contact with the substrate. These are particularly noticeable in squash rind and maltose agars in the tough layer of pink growth closely adherent to the agar (FIG. 8).

ASCOCARPS. Long-beaked, flask-shaped sexual fruits have developed only in very moist cultures at low temperature (FIGS. 1, 2, 3).

Sterile water dropped by pipette onto the surface of the mycelium is often necessary to stimulate their formation. At their optimum temperature of 15° C. they mature in from six to ten days. They develop more readily in cultures kept in a cold room, with slightly varying day and night temperatures and with alternating daylight and darkness, than they do in cultures kept in a low temperature incubator. This is perhaps due to the increase in the surface-droplets of moisture naturally developing with changing temperature. The ascocarps may be scattered, or clustered densely if water has stood on the substrate or on the mycelium. These structures are usually erect, though they may assume various angles with relation to the substrate, sometimes even lying prone, and buried in the agar (FIG. 3).

The ascocarp is pink, modified to a salmon or rusty shade. "Onion-skin-pink" on plate 28 of Ridgway's (1912) color standards is perhaps the correct shade. The length of the entire ascocarp on fruit rind cultures is 1 to 1.5 or often 2 mm.; occasionally extreme variations occur. The length of the beak is usually five to eight times the diameter of the basal bulb. The beak is tipped with from eight to fifteen spreading setae of varying length, the "crown of eyelashes" of Münch (FIG. 11). The soft bulbous base is composed of pseudoparenchymatous tissue; the inner layers break down to form a gelatinous matrix in which the spores mature (FIG. 19). The mature asci are softly angulated, with a thick gelatinous wall. They are eight spored, evanescent, and disappear soon after the spores are delimited (FIG. 12). The ascospores are oval and usually biguttulate, commonly $4.4 \times 8.8\,\mu$ (FIG. 13). Their discharge through the long beak is preceded by the appearance of a clear globule of liquid resting on the setae. If a current of air strikes this droplet before the spores have been discharged into it, the liquid is withdrawn into the interior, to be extruded again later. The spores collect in this terminal drop as they are ejected, making it milky white. At this stage the appearance of a thickly fruited culture is as of many pearls borne on delicate pink pedestals. As the spore clusters dry they become cream or tan colored, but the spores never lose their hyaline character. Often conidia are budded terminally from the setae (FIG. 11), and germinate in situ. the hyphae growing downward to neighboring



FIGS. 9–19. *Sphaeronaemella fimicola.*

ascocarps or to the mycelium, forming delicate festoons, quite characteristic of older culture plates. Within the ascocarp, after the discharge of the spores to the setae, fine mycelial threads grow through the wall of the ascocarp into the gelatinous matrix which may still exist in the basal bulb. These wandering hyphae, together with the absence of the classical club-shaped asci, and the presence of some lingering hyaline oval ascospores in the perithecial cavity, create the distinct illusion of a pycnidial rather than of an ascigerous structure. If these intruding branching hyphae are analogous to paraphyses, they do not resemble them in general appearance.

HETEROTHALLISM. Removal of the slime droplet on the setae to sterile water eventually frees the spores for single spore isolations. Cultures obtained from these single spores are far less vigorous than those from multiple-spore inocula. This species is heterothallic. No ascocarps have ever developed from any of the many single ascospore strains which have been obtained. When such single spore strains, complementary in nature, are crossed it has usually been found necessary to place a large mycelial inoculum from each of the two strains in very close proximity on an agar plate in order to secure perithecia. The physiological explanation has not been determined. Monoascospore strains seem more prone to morphological variation than multiple-spore strains, which fact may have an important bearing on the problem.

HABIT. Four collections of this species have been made from rotting cucurbits from New Jersey and Connecticut, each time after a "wet" summer. Thus far it has not been found in the field after a season of diminished rainfall. It was first noticed in moist chamber preparations of hubbard squash heavily infected with *Mycosphaerella citrullina* (Smith) Grossman; the pink beaks of the ascocarps of our mold rose above a thick expanse of the pink conidia of *Mycosphaerella*. Subsequent to study in pure culture, the characteristic snow white mycelium with its long chains of conidia was later recognized in the field against the dark rind of acorn squash. Mites (*Tarsenema*) are so fond of this fungus that it was some months before a sufficiently exact technique for their exclusion was developed. In cultures to which they at first gained access no ascocarps matured; the initial cells were devoured by the

intruders. The prevalence of such mites on rotting fruits could be one reason for the failure to find ascocarps developing in the field.

PATHOGENICITY. Repeated attempts to grow this fungus on various cucurbit fruits and on fresh twigs have consistently met with failure, though it grows well on the same fruits after they have been autoclaved. Its habit is essentially saprophytic.

SYNONYMS. A search of the literature has yielded one very brief description of a similar mold collected in North America.[1] Ellis and Everhart (**1889**) described *Sphaeronaemella carnea* as a new species, collected seventeen years earlier from ash bark at Lake Skaneateles, N. Y. We have not been successful in locating any type material of this species. As a check, autoclaved moist twigs (elm) were inoculated with our cultures. These twigs supported good growth of pink perithecia.

Jaczewski (**1898**) in his monograph on *Sphaeronaema* describes two species which resemble ours: *Sphaeronaema fimicola* (*Sphaeronamella fimicola* Marchal) and *Sphaeronaema Helvellae* (*Sphaeria Helvellae* Karsten; *Sphaeronaemella Helvellae* Marchal). Through the courtesy of the late Dr. Linder it was possible to make a direct comparison with some of Karsten's original *Sphaeria Helvellae,* Fungi Fennic. no. 674, in the Farlow Herbarium. Our fungus is definitely not the same species. No type material of *Sphaeronaemella fimicola* or of *Sphaeronaema fimicola* could be located either here or abroad (two or three years before the last war), but the Farlow Herbarium has one of von Höhnel's slides made from material collected by Keissler in Vienna, S. u. 1185. The specimen is well preserved, and in color, general appearance, and measurements agrees with our collection. Moreover, our species checks well with Marchal's (**1891**) rather full original report, and with the description and figures of *Sphaeronaemella fimicola* by Massee and Salmon (**1902**). These authors repeatedly recovered their species from dung of rabbit, fox, deer, and dog. As a check again, our

[1] Grateful acknowledgment is made of the generous assistance given by Dr. C. L. Shear, Dr. S. F. Ashby, Dr. W. Romeyn, Dr. Walter Snell, Dr. Fred J. Seaver, Dr. John A. Stevenson and the late Dr. David Linder, in the attempt to locate type material.

species was tested on like substrate, and grew vigorously on moist sterile rabbit dung.

In the absence of type material and of a fuller description of *Sphaeronaemella carnea* our fungus becomes *Sphaeronaemella fimicola* (Marchal) emend., with the following diagnosis.[2]

SPHAERONAEMELLA FIMICOLA (Marchal) emend.

Sphaeria fimicola Marchal.
Sphaeronaemella fimicola Marchal. Soc. R. d. Bot. d. Belgique, Bull. **30**: 143. 1891.
Sphaeronaema fimicola Jaczewski. Soc. d. nat. a. Moscou, Nouv. Mem. **15**: 300. 1898.
Sphaeronaemella carnea E. & E. Jour. Myc. **5–6**: 152. 1889–1891.

Fungo conidiophoro: Mycelio hyalino, pubescente; hyphis 1.2–3.5 μ diam., rare usque ad 5.6 μ; conidiis modo *Cephalosporii* et *Verticilli* praeditis atque catenis latis; magnis sporis perdurantibus in mycelio fictis circum perithecium basi, 5.6–16 μ, glabris vel verrucosis.

Fungo ascophoro: Mycelio heterothallico. Peritheciis superficialibus vel parum immersis, semper ferme gregariis sed aliquando sparsis, sphaeroideis vel ovatis, 150–250 μ diam., mollibus, luteolis vel carnosis coloribus, semper ferme glabris; in ostiolum, subulatum cylindraceo-conicum, vertice hyalino penicillato, 700–1800 μ, longitudine productis; sporulis ellipsoideis, hyalinis, leniter curvatis vel rectis, 7–8.8 μ rare 4.4–11.4 μ longis, 2–4.5 μ latis; muco obvolutis, ad ostiolo apicem demum expulsis et globulum ovoideum albidum formantibus.

Hab.: in fimo leporis usque canis, Belgium, 1883–1885; in fimo leporis usque cervae, England, 1901; in cortico Cucurbitae, New Jersey usque Connecticut, U. S. A. 1934–1945.

Specimens of this fungus are being deposited with the New York Botanical Garden, the Farlow Herbarium, and the Cornell University Herbarium. Cultures have proven so susceptible to temperature that they could be maintained only during the five years when a low temperature incubator (free from mites) was available, and therefore cannot be distributed at the present time.

SUMMARY. A species of fungus previously described as *Sphaeronaema fimicola* is reported from cucurbits in New Jersey and Connecticut. The supposed pycnidia have proved to be ascocarps, and the species is diagnosed as *Sphaeronaemella fimicola* (Marchal) emend. This mold is heterothallic, and its characteristics in culture are described and figured.

[2] We are indebted to Mrs. Vivian Trombetta Walker for latinizing the species diagnosis.

DESCRIPTION OF FIGURES

FIG. 1, Mature ascocarps on ten day old agar plate. × 8. 2, Detail of same ascocarps. × 14. 3, Young ascocarps submerged in the same agar plate. × 14. 4, Tufted mycelium of a monoascospore strain after fourteen days on a sterilized piece of gray Hubbard squash rind. × about 4. 5, Mycelium of monoascospore strain after ten days on Hubbard squash rind agar. × about 4. 6, Fan-shaped branches of mycelium from a monoascospore strain after four weeks of growth on zuccini squash rind agar. × 28. 7, Conidial masses in the mycelium of a ten day old squash rind agar culture made from mixed ascospore inoculum. × 68. 8, Base of an eight months old squash rind agar slant showing the tough moist pink under surface in the region where yeast-like cells are often abundant. × 1½.

FIG. 9. Free hand drawing of tufts of conidiophores at margin of three weeks old colony on sterilized squash rind. × about 25. 10,* Mature ascocarp from four weeks old cornmeal agar plate. The spore globule at the ostiole has dried, and slipped to one side of the beak. This spore globule is often found far down the side of the beak on an old dried ascocarp. × 43. 11, Tip of beak showing setae, with conidia developing at *a*. From 10 day old squash rind culture. × 660. 12, Asci from a perithecium in which were many spores already freed by the deliquescing of the thick ascus wall. × 660. 13, Ascospores from globules at tip of beak; *a*, mucilaginous matrix which becomes vacuolated in a water mount, and slowly breaks up; *b*, globules of matrix adhering to spore; *c*, hypha from spore germinated in the globule. × 66. 14, One of a cluster of bristle-like branches developing on the original bit of inoculum on a seven months old malt extract agar slant. × 480. 15, Chlamydospores from mycelium at base of ascocarp in six weeks old squash rind agar plate, showing both rough and smooth walled cells; *a*, germinating spore. × 660. 16–18, Conidia. × 660. 16, From nine day old squash rind agar plate, kept very moist with water well. Budding was active, and detached buds were frequently found. 17, From submerged portion of a ten day old squash rind culture. 18, From aerial conidiophores of same culture. 19, Portion of a section through a young ascocarp, with the outer wall omitted; asci in very early stages of development are shown among the gelatinizing pseudoparenchymatous cells. Section 5 μ thick; iron-alum haematoxylin. × 660.

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* 10–19. These drawings were made at table level with the aid of a camera lucida.

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NOTES AND BRIEF ARTICLES

HEMITRICHIA VS. HYPORHAMMA: The genus *Hemitrichia* was established by Rostafinski (Versuch 14. **1873**) to include a group of species of Myxomycetes, previously included for the most part in *Trichia,* characterized by a capillitium in which the spirally-marked elaters are united into a net rather than being free as in the species retained in *Trichia.* The representative species designated were *T. clavata* Pers., which may be regarded as the type of the genus, and *T. serpula* (Scop.) Fries. Two years later (Monog. p. 261. **1875**) Rostafinski arbitrarily changed the generic name to *Hemiarcyria* and the species involved were generally referred to that genus until Lister (Mycetozoa, p. 174. **1894**) revived the earlier name, since which time *Hemitrichia* has been universally used.

The distinction between *Trichia* and *Hemitrichia* is somewhat artificial. Certain species of *Hemitrichia, H. Vesparium* for example, have relatively free elaters and some species of *Trichia* may under certain conditions develop a more or less netted capillitium, but on the whole the distinction is useful and workable.

Some years before Rostafinski published, Corda (Icones, **6**: 13, *pl. 2, f. 34.* **1854**) founded the genus *Hyporhamma* upon *Trichia reticulata* Pers. (Syn. Fung. p. 182. **1801**), calling it *H. reticulatum* and making it the sole representative of his new genus. This species is universally recognized as identical with what is currently known as *Hemitrichia serpula* (Scop.) Rost. Corda's species is cited by Rostafinski (Monog. p. 266. **1875**) as a synonym of his *Hemiarcyria serpula,* although the volume and date of Corda's work are incorrectly given. In his generic diagnosis, Corda stresses the repent, *i.e.,* plasmodiocarpous, character of the fructification and the keel-like base of the peridium as well as the netted capillitium. The first character is true only of *H. serpula* and, in much less degree, of *H. Karstenii.* The keeled base of the peridium is present only in occasional specimens of *H. serpula.* Reference to the illustration shows the spirally-marked capillitium

and leaves no doubt that Corda actually had before him a specimen of *H. serpula*. But the textual comment as well as the drawing makes it clear that the keel-like base which Corda stressed was actually due to the fruiting of the plasmodiocarp along the surface of a rootlet, or, more probably, a rhizomorph of some fungus occurring superficially on the substratum, which Corda regarded as belonging to the myxomycete, stating that the keel develops from the root-like strands. He evidently regarded this character as of primary importance, since he selected a name for his genus based upon it, *'υπο + 'ραμμα* clearly referring to the basal thread.

To accept Corda's genus would involve a new combination in every species of *Hemitrichia* recognized at the present time, including the type species, since the specific epithet adopted by Corda is untenable, and would also necessitate radical emendation of the genus, because the repent habit and the carinate base are wholly inapplicable to the great majority of the species. Under such circumstances, if it were necessary, a good case could be made for conservation of *Hemitrichia* against *Hyporhamma*. Inasmuch, however, as Corda's genus was evidently based upon two discordant elements, the plasmodiocarp of the myxomycete and the rootlet or rhizomorph on which it was fruiting, it seems permissible, despite the fact that the species described is clearly recognizable, to regard *Hyporhamma* as technically a *nomen confusum* under the provisions of Art. 64 of the International Rules and advisable to publish this justification for the retention of the current and universally recognized name *Hemitrichia*.—G. W. MARTIN, Department of Botany, University of Iowa, Iowa City.

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